Aquatic Ecosystems Field Sampling Protocols

Alberta
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GLOSSARY

The following terms and acronyms are used in the document:

Ambient	natural, existing or baseline information
ATC	air temperature compensation
AWOS	Automated Weather Observation System
Biofilm	thin organic film composed of a mix of organisms (algae, fungi, bacteria) and their secretions
BTEX	benzene, toluene, ethylbenzene, and xylene
CAEAL	Canadian Association of Environmental Laboratories
cfs of ft^3/s	cubic feet per second
cms or m^3/s	cubic metres per second
COCs	chain of custody forms
Conductivity	a measure of the ability of a solution to carry an electrical current. Conductivity is dependent on the total concentration of ionized substances dissolved in the water and is measured as microsiemens per centimetre (μS/cm).
DDW	double deionised water
DO	dissolved oxygen – Concentration of oxygen dissolved in water, where saturation is the maximum amount of oxygen that can theoretically be dissolved in water at a given altitude and temperature. Expressed as milligrams per litre or as percent saturation.
ERP	Emergency Response Plan
Field Work	that component limited to activities conducted out of the office
Flow	volume of water passing a given point per unit of time. The units of measurement are typically either cubic metres per second (cms or m^3/s) or cubic feet per second (cfs or ft^3/s). Synonymous with discharge.
GIS	Geographic Information Systems – Systems for identifying locations geographically and organizing information about those locations in a relational process based on shared geographic location. Data are referenced with geographic coordinates and stored in digital format in a computer.
GPS	Global Positioning System – System of satellites in permanent orbit above the earth that allow a receiver to triangulate their position on or above the earth's surface.

Groundwater	subsurface water in a zone of saturation, standing in or passing through (groundwater flow) the soil and the underlying strata.
IBW	Inorganic-grade Blank Water
JSA	job safety analysis
Littoral	region of shallow water where light reaches the bottom.
masl	metres above sea level
Meiofauna	aquatic animal life that pass through 500 µm screen but are retained by a 64 µm screen
Method	a regular, systematic or orderly way of conducting field sampling; a step by step procedure
Mixing	internal circulation in a water body.
Mixing depth	depth of body of water where mixing occurs.
Mixing zone	area of location of a water body where individual masses of water are mixed.
NTU	nephelometric turbidity units
PADI	Professional Association of Diving Instructors
PBW	Pesticide-grade Blank Water
Periphyton	an assemblage of small plant organisms (mostly algae) that are firmly attached to solid surfaces underwater (also referred to as Benthic Algae)
PFD	personal floatation device
pH	measure of the acidity and alkalinity of a solution, expressed as negative $\log_{10}$ of the hydrogen-ion concentration on a scale of 0 (highly acidic) to 14 (highly basic). A pH of 7 is neutral.
POC	particulate organic carbon
PPE	personal protective equipment
Protocol	a plan detailing the key elements of a procedure; a guidance document
RAMP	Regional Aquatics Monitoring Program
RST	reference steel tape
SCC	Standards Council of Canada
Specific Conductance	a measure of the ability of a solution to carry an electrical current, similar to conductivity, but compensated to a temperature of 25°C.
Surface Water	standing water above the substrate or water that flows exclusively across a land surface and includes all perennial and ephemeral water bodies.

TDG	Transportation of Dangerous Goods
TEH	total extractable hydrocarbons
TOC/DOC	total or dissolved organic carbon
TPC	total particulate carbon
Turbidity	refers to the relative clarity of a water body; a measure of the extent to which light penetration in water is reduced from suspended materials such as clay, mud, organic matter, color or plankton. The units of measure are by several nonequivalent standards such as nephelometric turbidity units (NTU), formazin turbidity units (FTU) and Jackson turbidity units (JTU).
TVH	total volatile hydrocarbons
UTM	Universal Transverse Mercator – Grid system for establishing a fixed point between 84°N and 80°S using exact measurements.
Velocity	distance traveled per unit time
VPP	Volatile Priority Pollutants
Wetted Width	the width of the water way that is occupied by water

1.0 INTRODUCTION

This document is one of a series of four reports containing field protocols and methods for the sampling of key components of aquatic ecosystems in Alberta (i.e., for surface water quality, surface water quantity, groundwater, and meteorology). These protocols are important to ensure that samples are collected consistently and the data obtained are accurate and scientifically sound. Given appropriate sampling protocols and study designs, the information obtained will permit the tracking of changes in surface water quality and other measures of ecosystem health over time. This will also allow for accurate comparisons of ecosystem health among different watersheds and ecoregions in Alberta.

This document is a revision and update to the field manual, "*Water Quality Sampling Methods*" (Alberta Environment 2002) that outlines the procedures used by Alberta Environment. The manual was reviewed for gaps and redundancies. The deficiencies were addressed with relevant information from protocols used by other agencies including Alberta Agriculture, Food and Rural Development, Alberta Sustainable Resource Development, British Columbia Ministry of Water, Land and Air Protection, Ontario Ministry of the Environment, Manitoba Conservation, Environment Canada, Department of Fisheries and Oceans and United States Environmental Protection Agency. The final protocols in this document are considered as best practices for field sampling in aquatic ecosystems of Alberta. Each protocol consists of a brief description of purpose, general background on the sampling technique, the type of sampling equipment required, a step-by-step procedure to collect and handle the sample, and a reference section for further information.

It should be noted that the sampling manual focuses on field sampling procedures and it does not cover the sampling design of field programs. Also, the manual does not represent a comprehensive presentation of needs related to quality assurance/quality control (QA/QC), data validation and safety considerations related to field sampling. Further information on QA/QC and safety issues can be found in a variety of sources such as, BC-RISC (1997 a and b), USEPA (2000b), USEPA (2002), Environment Canada (2002, 2004), BC-MWLAP (2003), RAMP (2005), USGS (2005), Alberta Environment (2005), and Mitchell (2006).

2.0 LAKE AND RESERVOIR PROTOCOLS

2.1 Multiprobe Field Measurements

Purpose

In situ measurements of parameters such as pH, dissolved oxygen, temperature, conductivity, turbidity, and redox potential (when required) are routinely taken at the time of sampling in lakes and reservoirs. These measurements are taken *in situ,* in the water body just below the surface, at mid-depth or at discrete depths depending on the sampling objective and the depth of the sampling site, using electronic single or multiprobe meters. Some water quality parameters should only be measured *in situ* (i.e., temperature) or the measurement (e.g., pH, turbidity) could be influenced by transportation time for samples to the laboratory.

General

Taking accurate *in situ* measurements depends on strict adherence to calibration, maintenance and QA/QC procedures for the electronic water quality meters used. Maintenance and calibration should be carried out according to the manufacturer instructions and additional technical assistance can be found on the internet and through consultation with technical representatives. The manufacturer manuals are not necessarily comprehensive, so an additional information compilation exercise through these sources is often worthwhile. Maintenance and calibration logs should be kept up to date to track the performance of the meter (see Appendix A). The meter probes should be calibrated daily under field conditions and temperatures, and periodically throughout the day if required (e.g., dissolved oxygen at sites of different altitudes or every five samples if water quality changes dramatically from site to site). The exceptions are: temperature (check in lab monthly with a mercury thermometer), conductivity and turbidity (calibrate at the beginning of the sampling trip), and redox potential (calibrate once every six months). Always calibrate the pH meter with at least two buffer solutions (either pH 7 and pH 4 (sample pH <7) or pH 10 and pH 7 (sample pH >7)). Consult the manufacturer manuals and available information for the meter for specific calibration information.

Review the water quality data on-site during sample collection to prevent the measurement and/or recording of false measurements. Re-measure and double-check any dubious readings before leaving the site. It may also be worthwhile to verify data at the end of the day for some key parameters (not re-calibrations) to check if the meter has drifted or is malfunctioning. Meter readings should be checked in standard solutions and recorded in the log/field book. This ensures that the meter has been working properly throughout the day. It is preferred to take field water quality readings from the body of water itself (*in situ*) but on some occasions it may be necessary to take the measurements from a sub-sample of water. In this case take separate water samples for these field measurements and never take field water quality measurements from samples to be submitted to the laboratory for analysis.

Equipment

- Single or multiprobe electronic water quality meters from reputable brands (e.g. YSI®, Hydrolab®, etc). The recommended minimum accuracy for the field measurements is: DO (± 0.2 mg/L); temperature (± 0.4 °C); conductivity (± 0.05 µS/cm); pH (± 0.02 units); turbidity (± 0.01 NTU); and salinity (± 0.1 ppt).
- Standard calibration solutions (within the expiry date).
- Field log book and calibration log sheet/book (waterproof paper).
- Lint free wipes (e.g., Kimwipes®), tap water, distilled water.
- Barometer and thermometer.
- Winkler equipment (see Section 3.2).
- Pelican® Case to store the meter when not in use.
- The meter manual and any items required to change membranes or perform minor multiprobe repairs.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals (e.g., calibration solutions)

Procedure

1. Calibrate the multiprobe/meter prior to daily sampling for pH and DO. Conductivity and turbidity can be calibrated at the beginning of the sampling trip and redox potential should be calibrated every six months. Temperature should be calibrated every month with a standard thermometer.
2. *In situ* measurements at sites >4 m deep, should be taken just below the surface of the water (0.1 m depth) and at 1 m intervals down to 1 m above the lake bottom. At sites ≤2 m deep, one set of measurements at mid-depth might be considered appropriate. At sites between 2 and 4 m deep, two measurements can be taken 0.25 m below the surface and 0.25 m above the lake bottom.
3. Let the instrument stabilize at each depth (usually 1-2 min) and record the readings in a field sheet/book (an example of a lake profile data sheet is in Appendix B). Also, if possible store readings at each depth in a datalogger. On deep profiles and where approved by the project manager, it may be acceptable to proceed at 5 m intervals when there is little change in readings at 1 m intervals. When change is detected (thermocline, chemocline etc.), then define the area of change at 1 m intervals.
4. Bring the probe/sonde back up to 1 m, allow it to stabilize and record readings at that depth. (Note: redox will probably not stabilize quickly at the surface.) This acts as a field check on the instrument and verifies the accuracy of the first reading.
5. One water sample taken at one profile depth per water body is subjected to a Winkler analysis as a further check of the accuracy of the meter measurement, preferably at a depth where oxygen appears stable. A meter DO measurement within ±0.5 mg/L of the Winkler DO measurement is generally considered acceptable, however USGS (2005) recommended that meter and Winkler DO measurements should be within ± 0.05 mg/L DO.

Reference

This protocol was derived from Environment Canada (1983), Alberta Environment (2002), Environment Canada (2004) RAMP (2005), USGS (2005), and EMAN-North (2005).

Further information regarding multiprobe water quality measurements can be found in USGS (2005).

2.2 Secchi Disk Use

Purpose

A Secchi disk is a 20 cm diameter, flat disk with alternating black and white quarter-circles. It is used to provide a visual measure of water clarity or optical depth, by lowering it into the water and determining the depth where the disk disappears from view.

General

Take the Secchi depth measurement in the shade and do not wear sunglasses. Record the time of sampling as this can potentially affect the Secchi measurement. The optimal time for taking a Secchi measurement is mid-day. Take at least two measurements at each sampling site/station and estimate the optical depth based on the mean of these two measurements. Ideally the Secchi disk measurement should be accurate to within ± 1 cm. The higher the Secchi disk reading, the clearer the lake. If the light meter is not operational, determine the approximate euphotic zone (depth of 1% incident radiation [light]) by multiplying the Secchi disk reading by 2.

Equipment

- Secchi disk
- Calibrated cord (or in some cases a chain) attached to the disk - Preshrink the cord by soaking and drying it before marking the desired depth intervals, using a permanent marker. Refrain from using tape as a marker because it can loosen and move on the cord over time.

Procedure

1. Take the Secchi reading on the shaded side of the boat.
2. Slowly lower the disk into the water until it disappears from sight and note the depth (Depth 1).
3. Lower the disk down a further 1 m (or until it is well out of sight) then slowly raise the disk until it is visible again and note this depth (Depth 2).
4. The Secchi disk reading is the average of the two recorded depths (Depths 1 and 2). Record the time of sampling.

Reference

This protocol was derived from Environment Canada (1983), Alberta Environment (2002), Environment Canada (2004), RAMP (2005) and EMAN (2005).

2.3 Photosynthetically Active Radiation (PAR) Sensing Procedures

Purpose

PAR is a slightly narrower band of radiation (400 – 700 nm) than visible light, and it is that area of the spectrum used by plants. The measurement of PAR profiles using, for example, the LI-COR® Solar Radiation Measuring System, is undertaken to measure PAR attenuation with water depth. The euphotic zone extends to the depth where the PAR meter records 1% of subsurface incident radiation. We are mostly concerned with measuring the down welling radiation.

General

- If using the LI-COR® LI-1000 datalogger, stabilized readings are easier to choose if a 5 second averaging function is used. This has been programmed into the LI-COR® LI-1000.
- Readings should be taken under consistent light conditions. Note the sun or cloud conditions in the lake field sheet/book.
- Carry spare batteries for the datalogger/display.
- In rough weather, take the 2.5 cm readings just below the trough of the waves.
- If the lake is exceptionally green, stained or turbid with a Secchi less than 1 m, take extra light readings at 0.5 m intervals to aid in accurate calculation of extinction coefficients.
- Compare LI-COR® readings with another LI-COR® periodically throughout the season.
- The LI-COR® LI-189 display has no data storage capacity.
- Sensors should be rinsed with distilled/deionised/RO water periodically throughout the season.
- Sensors are periodically sent in to the factory for calibration (e.g. LI-COR® every two years).

Equipment

LI-COR® Solar Radiation Measuring System with Datalogger display LI-1000 or LI-189 display and LI-192SA underwater sensor. The light meter should be able to simultaneously measure light above and below the surface (i.e., two light cells).

Procedure

1. Read the manual and ensure the datalogger/display has the proper sensor calibration multiplier stored for the sensor in use.
2. Ensure all connections are properly made, remove protective cap from sensor and turn instrument on.
3. Take light readings from sunny side of boat and leave sensor at each depth for at least 15 sec or until stable.
4. Record and store readings at 2.5 cm, 10 cm and 1 m intervals to 1 m below the euphotic zone depth (1% of the 2.5 cm reading). If the initial readings drop by more than 50%, use 0.5 m intervals. Readings are in µm/sec/m^2.
5. Accurately determine the euphotic zone depth by raising the sensor to 1% of the 2.5 cm reading. Record this depth and sensor number in the lake field sheet.
6. Retrieve sensor and carefully repack in the storage case - do not kink the cable.

7. Transfer stored data to disk using a communications package.

Reference

This protocol was derived from Alberta Environment (2002).

2.4 Discrete Profile Lake Water Sampling

Purpose

Discrete profile sampling refers to the collection of water samples from specific points in a depth profile at a site in a lake or reservoir. Samples are typically taken from the deepest area of the water body. This type of sampling allows the characterisation of water quality at various depths, providing information on how water quality changes with depth. This may be influenced by a variety of factors such as layering or stratification of physical and chemical variables,. If contamination is an issue of concern in a waterbody, the water samples should be taken at sites progressing from the least contaminated areas to the most contaminated site last. The degree of contamination may be estimated from historical data, site conditions, land use, professional knowledge etc.). Two of the most commonly used methods to sample water at depth in lakes and reservoirs are: pumping water to the surface (e.g., using GeoPump® Sampler or Master Flex® pumps) and collecting water samples at depth (Van Dorn/Kemmerer Depth Samplers).

General

- Only use sample bottles provided by the analytical laboratory specific to each analysis. Reject any uncapped bottles (especially those for analysis of trace metals and other contaminants). Ensure there is always at least one extra set of bottles available.
- Ensure bottles remain capped until sample collection and are stored under clean conditions (e.g., in cooler, plastic bag etc). Vehicles should also be kept reasonably clean to limit potential contaminations.
- Only leave the sample bottle uncapped while filling the bottle and/or adding preservatives. Do not touch the cap liner or the inside of the sampling bottles (even while wearing gloves). Only the water sample and the preservative should touch the inside of the sampling bottle or the cap.
- Review documentation that accompanies the sample bottles which refers to sample collection, storage and transport, and consult with laboratory personnel regarding these requirements. If samples are to be submitted close to or on the weekend, make arrangements with the laboratory to ensure sample holding times are respected.
- Ensure all preservatives are sealed and within the marked expiry date. Add preservatives to samples in an area away from potential sources of contamination (e.g., roads and car parks [dust and hydrocarbons]). Read the MSDS sheets for all preservative chemicals and wear safety glasses and gloves while preserving the samples.
- Deep water sites to be sampled routinely throughout the year should be noted with GPS coordinates.
- Boats should always be anchored when doing a profile. Samples should always be collected at the bow of the boat. The bow will always point into the wind when anchored, reducing the potential for contamination from the boat or motor.
- Colour code samples bottles to ease collection and prevent mix-ups.
- Keep all sample collection and filtering apparatus in sealed clean plastic bags and/or in a clean cooler when not in use. This prevents contamination.
- Sampling personnel should wear unpowdered latex or polyethylene disposable gloves or long sleeved rubber gloves while collecting water samples. Refrain from smoking or

eating. Be careful with insect repellents and sunscreen and be certain none of it enters samples.

- Take a photograph and GPS coordinates at each site and follow procedures outlined in Section 2.1 for taking *in situ* water quality measurements. Record observations at the sampling site as well as the whole lake (e.g., algal blooms, water colour, macrophytes).
- See Section 4.2 for specific guidelines related to trace organic sampling in lakes.
- See Section 4.3 for sampling equipment cleaning and decontamination procedures.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

Peristaltic Pump

- Portable peristaltic sampling pump (GeoPump® or Master Flex®) with 12 volt battery, power cord and fuses. Always carry spare silicon pump head tubing and the 12 volt power cord. Test the pump battery charge before going in the field.
- Short pump tubing (outlet), 3/8" ID x 5/8" OD x 1/8" wall Tygon® or equivalent
- Silicon pump head tubing, sizing specific to pump being used with appropriate sized tubing connectors.
- Long pump tubing permanently marked in meters (inlet), 3/8 ID x 5/8 OD x 1/8 wall Tygon® or equivalent with stainless steel weight/intake on end.
- Van Dorn or Kemmerer depth sampler (for back-up in case of pump problems).
- Watch, waterproof field sheets/book and pens, labelling tape.
- GPS unit, depth sounder, camera.
- Sufficient sample bottles and preservatives for the number of depths to be sampled plus an extra set.
- Coolers and ice packs (do not use cube or block ice however if you do be very sure caps are on tightly, or are in Ziploc® bags) or hot water bottles, depending on the season.
- Lab analysis request sheets and chain of custody forms (COCs).
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Equipment for field cleaning/decontamination procedures (see Section 4.3)
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals

Van Dorn/Kemmerer Sampler

- 2-16 L Van Dorn/0.5-8.0 L Kemmerer samplers with messengers, and rope permanently marked in meters (sampler material must not interfere or compromise sample analysis).
- Van Dorn samplers are available in both horizontal and vertical configurations. Vertical configurations tend to be used in large lakes to ensure water is collected at the desired depths. The Van Dorn horizontal configuration samples a very narrow depth range and therefore is used to sample at, or just above/below a sharp thermocline, close to the lake bottom, or in very shallow lakes.

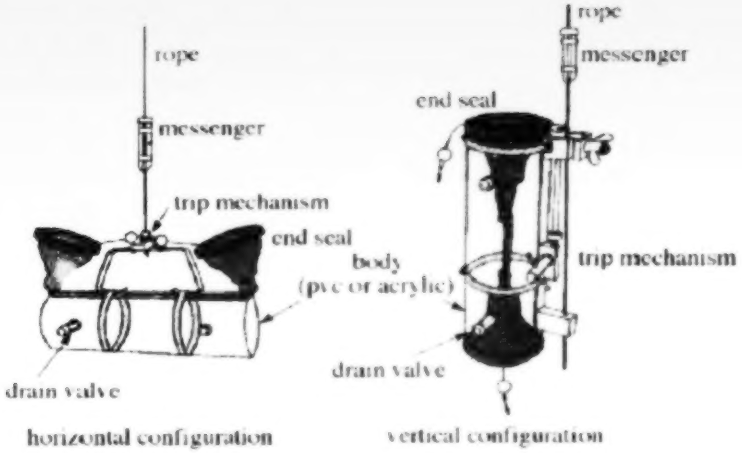
rope
messenger
trip mechanism
end seal
body
(pvc or acrylic)
drain valve
horizontal configuration
rope
messenger
end seal
trip mechanism
drain valve
vertical configuration

Figure 2.4-1 **Van Dorn depth water sampler – horizontal and vertical configurations (BC-MWLAP 2003)**

- Waterproof field sheets/book and pens, labelling tape.
- GPS unit, depth sounder.
- Sufficient sample bottles and preservatives for the number of depths to be sampled plus an extra set.
- Coolers and ice packs (do not use cube or block ice) or hot water bottles, depending on the season.
- Lab analysis request sheets or chain of custody forms (COCs).
- Equipment for field cleaning/decontamination procedures if needed for special sampling (see Section 4.3)
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals

Procedure

Peristaltic Pump

1. Attach the inlet and outlet tubing to the pump via connectors.
2. Lower the inlet tubing with the weight attached to the first sampling depth and run the pump for at least 5 min to flush the pumping system. Run longer if using long tubing to ensure tube is well flushed.
3. Note: Rinse all sample bottles and caps with water from the appropriate depth three times before filling them, except for those destined for:
    - bacteriological analysis, cyanide, synthetic organic compounds, pesticides;
    - volatile hydrocarbons (e.g. including total volatile hydrocarbons [TVH], total extractable hydrocarbons [TEH], benzene, toluene, ethylbenzene, and xylene [BTEX]; and
    - total petroleum hydrocarbons (e.g., TVH, TEH, and BTEX), oil and grease.

It is recommended that sample bottles for these parameters should not be rinsed because some parameters can accumulate on the container during rinsing. Also in the case of bacteriological analysis, this is done to maintain sterile conditions. Some sampling manuals recommend that sample bottles for additional parameters also not be rinsed, i.e.:

- suspended solids, parameters associated with suspended solids; and
- trace metals (although some laboratories may still request that trace metal bottles be rinsed).

Check with the project manager and the analytical laboratory as to whether sample bottles subject to the analyses described should be rinsed.

4. Do not touch the sample bottles with the sampler tubing. Hold the sample bottle caps lid-down while filling the sample bottle. Fill sample bottles with water from the appropriate depth noting head space requirements and other requirements specific to the parameter to be analysed. Typically sample bottles should be filled to approximately 0.5 cm from the top unless the laboratory has instructed that no headspace be left (e.g., volatile hydrocarbons) unless room is need for adding of preservative. Consult documentation provided by the analytical laboratory and laboratory personnel.
5. If ultra-clean techniques (e.g., "clean hands/dirty hands") are required to collect samples for some ultra-low level analyses then sampling/handling instructions provided by the laboratory should be followed (e.g., mercury).
6. Lower the inlet tubing to the next sampling depth - run the pump 1 min for each 10 m of tubing, before filling bottles.
7. When filling Winkler DO bottles place the outlet tube at the bottom of the bottle and allow three times the bottle volume to pass. Slowly remove the tube to prevent any aeration and stopper. Add 2 ml of manganous sulphate, and 2 ml of alkaline iodide azide, in quick succession then stopper it. Invert rapidly several times. Immediately repeat for the second bottle. Invert both bottles fifteen times then place a protective plastic cap over each stopper. Resample if large bubbles are present in the bottles. Keep the samples at 4 °C, in the dark, and titrate within 24 hours. Normally, the water quality meter DO value should be within ± 0.5 mg of the Winkler DO measurement.
8. Immediately after collection store the water samples at 4 °C in a closed cooler. Do not allow them to freeze. Preserve the previous samples while waiting for the tubing to flush.
9. Collect samples at prescribed intervals down to 1 m above the bottom.
10. After all depths have been sampled, raise the pump intake hose above the lake surface and run the pump until the tubing is empty.
11. Shut the pump off and store properly.
12. Filter and/or add preservatives to appropriate bottles, immediately, or as soon as possible after sample collection. If the samples are to be laboratory-filtered ship them as soon as possible and ensure they arrive at the laboratory well within the hold time specified for un-filtered and un-preserved samples. Unfiltered chlorophyll-a samples should be stored in a dark bottle to minimize light exposure.
13. All sample bottles should be clearly labelled with date, location, site, depth, analytical parameter group and sampler ID. Store and transport all samples at 4 °C in a closed cooler. Do not allow to freeze.

14. At the end of each sample day rinse the tube with tap water and fill with 5% HCl for 6-24 hours and then rinse thoroughly (at least three times) with distilled/deionised water (see Section 4.3 for detailed cleaning protocols).

Van Dorn/Kemmerer Sampler

Only use Van Dorn and Kemmerer samplers for lake sampling sites >2 m in depth and >1 m in depth, respectively. Ensure that the Van Dorn/Kemmerer sampler is functioning properly. Do not touch the inside of the sampler body or end plug. Store the sampler in the open position in a clean place.

Note: Graduate sampler line starting from the mid-point of the sampler tube to obtain correct water sampling depths.

1. Set the sampler in the open position.
2. Rinse the sampler three times with lake surface water, ensuring the outlet hose is also rinsed.
3. Lower the sampler to the desired depth, drop the messenger and retrieve the sampler. Allow a small volume of water to pass through the outlet tube to flush the drain valve. This further reduces the possibility of contamination with water from the previous site(s).
4. Note: Do not rinse all sample bottles. See rinsing instructions given in Step 3 in the previous sampling protocol for the Geo-pump® sampler.
5. Hold the sample bottle caps lid-down while filling the sample bottle. If the ultra-clean techniques (e.g., "clean hands/dirty hands") are required to collect samples for some ultra-low level analyses then sampling/handling instructions provided by the laboratory should be followed (e.g., mercury).
6. If necessary lower the sampler to collect more water from the same depth, repeating the above steps. Pour off Winkler DO samples the first time a given depth is sampled to prevent future aeration.
7. When filling Winkler DO bottles place the outlet tube at the bottom of the bottle and allow three times the bottle volume to pass, slowly remove the tube to prevent any aeration and stopper. See further sampling, preservation and storage and instructions given in Step 7 in the previous sampling protocol for the Geo-pump® sampler.
8. Collect water samples at the required depths down to 1 m above the lake bottom, repeating the above steps. Always work from the top to the bottom of the water column. Immediately after collection store the water samples at 4 °C in a closed cooler. Do not allow to freeze.
9. Filter and/or add preservatives to the appropriate bottles, immediately, or as soon as possible after sample collection. If the samples are to be laboratory-filtered ship them as soon as possible and ensure they arrive in the laboratory well within the specified hold time for un-filtered and un-preserved samples.
10. All sample bottles should be clearly labelled with date, location, site, depth, analytical parameter group and sampler ID. Store and transport all samples at 4 °C in a closed cooler. Do not allow to freeze.

Reference

This protocol was derived from: Environment Canada (1983), BC-RISC (1997a), USEPA (2000a), Alberta Environment (2002), BC-RISC (2002), BC-MWLAP (2003), Environment Canada (2004), RAMP (2005), and EMAN-North (2005).

2.5 Composite Integrated Water Sampling

Purpose

The collection of a water quality sample across a depth range at a particular site is referred to as integrated sampling (e.g. spanning the euphotic zone). Consequently, composite integrated sampling refers to the collection of integrated water quality samples at several sites in a water body, which are then pooled to form a single composite integrated water quality sample for that water body. A composite integrated water quality sample accounts for horizontal and vertical spatial heterogeneity that may exist in that water body (i.e., provides an estimate of average water quality). The method provided here relates to the use of a sampling tube to collect the depth integrated water sample, however a Van Dorn or a Kemmerer depth sampler can also be used. If a depth sampler is used to collect a composite integrated sample, the sampling guidelines described in Section 2.4 for discrete depth sampling should be adapted for the collection of an integrated water sample.

General

- The number of sites and locations sampled will depend on the project (minimum of ten sites per lake) and should be discussed with the project manager prior to sampling. Sample ten sites in larger lakes with a euphotic zone >5 m, but sample fifteen sites in larger lakes with a euphotic zone <5 m.
- Calculate the required number of hauls per site needed to collect the total sample volume. This depends on the volume requirements of the variables being analyzed.
- The total sample volume = No. of hauls x No. of sites x Euphotic zone depth x the amount of water collected per meter of haul hose (approximately 0.1 L/m).
- The same number of hauls must be taken at each site.
- Between hauls, place the sample tube foot valve in a clean place.
- Avoid taking water in an area disturbed by the boat or floatplane.
- See Section 4.2 for specific guidelines related to trace organic sampling in lakes.
- See Section 4.3 for sampling equipment cleaning and decontamination procedures.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Sampling container - 10-20 L capacity, such as a large carboy, complete with lid and light proof container (or use a black garbage bag to cover more transparent containers).
- Two sampling tubes (1/2" ID x 3/4" OD x 1/8" wall Tygon® tubing or equivalent) of appropriate lengths (usually 5 m and 10 m long, depending on water clarity) with a 3/4" foot valve and weight on one end.
- Phytoplankton bottles (100-150 ml dark glass screw tops).
- Lugol's solution, freshly acidified, 3 ml/sample (see Appendix C).
- Hydrographic map, labelling tape and waterproof field sheets/book and pens.
- Long rubber gloves, polyethylene or non-powdered latex gloves.
- Sample bottles and preservatives in coolers plus an extra set of sample bottles and preservatives. Ice packs or hot water bottles, depending on the season.

- 10 L carboy of deionised water. Use fresh deionised water (do not store for extended periods of time; do not use after 6 months)
- Filtering equipment and supplies stored in sealed plastic bags (see Section 2.7).
- GPS unit, depth sounder, camera.
- Lab analysis request sheets and chain of custody forms (COCs).
- Equipment for field cleaning/decontamination procedures (see Section 4.3)
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals.

Procedure

1. Rinse the sampling tube, sample bucket and lid three times with lake water.
2. Place the sample bucket in a light proof container (or black plastic bag) to reduce light penetration and associated phytoplankton chlorophyll production. Place the open end of the sampling tube in the hole in the sampling bucket lid.
3. Lower the weighted end of the tube slowly (approx. 1 m per sec.) and vertically though the euphotic zone. If the depth of the sample site is less than the depth of the euphotic zone, only sample to within 1 m of the bottom.
4. Pull the tube up into the boat.
5. Check for sediment in the water sample before it leaves the tube.
6. If there is no sediment in the tube, invert the foot valve and drain the water into the sample bucket. If there is sediment in the tube, discard the sample, rinse the tube 3-5 times with lake water, move the boat a few meters, and repeat the haul to a shallower depth.
7. If any sediment is introduced to the sample bucket, discard the sample and start over. Ensure that the sampling tube and bucket are well rinsed (3-5 times).
8. Repeat steps 3-7 for all sites.
9. After all sites have been sampled, remove the sampling tube from the sample bucket.
10. At the end of each sampling day, rinse the tube and bucket with tap water. Remove all metal parts and immerse in or fill with 5% HCl for 6-24 hours, then rinse at least three times with distilled/deionised water (see Section 4.3 for detailed cleaning protocols).

Sub Sampling Procedures

1. Cap and shake the sample container well (approx. 30 sec.) before pouring each sample.
2. Minimize dust contamination by pouring samples in a calm area and by capping sample bottles immediately before and after filling. Do not touch the inside of the sample bucket, bottle lids or mouths with your hands.
3. Note: Do not rinse all types of sample bottles. See rinsing instructions given in Step 3 in the Peristaltic pump sampling protocol (Section 2.4).
4. Hold the sample bottle caps lid-down while filling the sample bottle. If the ultra-clean techniques (e.g., "clean hands/dirty hands") are required to collect samples for some ultra-low level analyses then sampling/handling instructions provided by the laboratory should be followed (e.g., mercury).
5. Immediately after collection store the water samples at 4 °C in a closed cooler. Do not allow to freeze.
6. Filter and/or add preservatives to the appropriate bottles, immediately, or as soon as possible after sample collection. If the samples are to be laboratory-filtered ship them as

soon as possible and ensure they arrive in the laboratory well within the specified hold time for un-filtered and un-preserved samples. Unfiltered chlorophyll-a samples should be stored in a dark bottle to minimize light exposure.

7. Add 3 ml of Lugol's solution to the 150 ml phytoplankton sample. Add 5-6 drops of buffered Formalin to the 150 ml phytoplankton samples, 3 to 24 hours after sampling to inhibit bacterial or fungal growth. CAUTION: 100% FORMALIN IS A SUSPECTED CARCINOGEN. READ THE MSDS.
8. All sample bottles should be clearly labelled with date, location, site, depth, analytical parameter group and sampler ID. Store and transport all samples at 4 °C in a closed cooler. Do not allow to freeze.

2.6 Spatial Composite Water Sampling

Spatial composite sampling refers to water quality samples taken from just below the surface (grab sample), or at a discrete depth from various spatial locations within a lake/reservoir, and then combined to form a composite sample. This type of composite sampling is another method to account for horizontal heterogeneity within a water body (i.e. provides an average estimate of water quality). This method is normally used on shallow lakes.

The Regional Aquatics Monitoring Program (RAMP) collects grab water samples from 30 cm below the water surface from five locations within a lake and combines them as described in Section 2.5 to form a spatial composite sample. The number of locations to be sampled within a lake should ultimately be determined by the project manager and may vary according to the lake size and study objectives, as noted in Section 2.5.

Where the spatial composite sample is comprised of discrete depth samples, the sampling procedures described in Section 2.4 for the Peristaltic pump and the Van Dorn and Kemmerer samplers should be used to take a discrete water sample at each location. The discrete water samples should then be pooled in a sampling container to form one composite sample. This sample should be sub-sampled as described in Section 2.5 for the various analytical variables.

Where the spatial composite sample is comprised of grab samples taken 30 cm below the water surface, the following procedure should be adopted for grab sampling at each location.

- A pre-cleaned intermediate sample bottle should be rinsed three times with lake water prior to final sample collection. Submerge to a depth of 30 cm, uncap and fill bottle, and recap at depth (to avoid contamination).
- At the surface, remove the lid and dispense away from the sampling site. Repeat this procedure twice more before filling the container at 30 cm below the surface and recapping at depth. While sampling avoid submerged vegetation and ensure sample is free of obvious foreign material not representative of the water column at time of sampling (e.g. algae, sediment, organic matter etc.).
- Pour the water sample into the composite sample bucket and continue to sample until sufficient composite volume has been collected (do not rinse again). Keep the bucket covered during sampling.

Reference

This protocol was derived from Environment Canada (1983), BC-RISC (1997a), USEPA (2000a), Alberta Environment (2002), BC-MWLAP (2003), Environment Canada (2004), RAMP (2005), and EMAN-North (2005).

2.7 Chlorophyll-a Sample Filtering

General

- Ensure that the sample bucket and filtering apparatus are kept out of direct sunlight.
- Rinse the filter funnel and graduated cylinder between samples with deionised water.
- Always use the forceps marked for chlorophyll-a filtering when handling filters.

Equipment – Field Filtering

- 500 ml and 1000 ml Erlenmeyer filter flasks
- 500 ml graduated cylinder
- 2 filter holders with No. 8 stoppers
- GF/C glass fibre filters 4.7 cm
- 0.45 um cellulose acetate filters 4.7 cm
- Forceps
- Masking tape, label tape
- Aluminum foil squares (10x10 cm)
- Pens, waterproof markers
- Saturated $MgCO_3$ solution/anhydrous $MgCO_3$
- Deionised water (10 L carboy)
- 2 hand vacuum pumps
- 2 squeeze bottles
- 90% acetone
- Chlorophyll tubes

Equipment - In-Line System

- 2 - 1000 ml flasks
- In-line filter holder
- 142 mm 0.45 Fm cellulose-acetate filters
- Variable speed peristaltic pump
- 9 L plastic tray

Procedure

1. Filter using only the apparatus and flask designated for chlorophyll-a (do not acid wash). Do not exceed 7 psi or 48 kPa when filtering. Triplicates are required.
2. Rinse the graduated cylinder with distilled/deionised water.
3. Mix the sample for 30 sec before pouring off first replicate.
4. Lakes: Filter 50 - 500 ml of sample through a 47 mm GF/C filter and record the volume on the lab sheet (use tweezers when handling the filter).
5. Rivers: Filter 500 - 1000 ml of sample through a GF/C filter and record the volume. If water is very turbid, filter what you can through two filters and combine in one tube. Note total volume and "two filters" on field sheet.
6. Filter just enough of a sample to get a light green/brown colour on the filter. Do not clog the filter.

7. Rinse the graduated cylinder and the sides of the filter funnel with distilled/deionised water and pass this rinse water through the filter.
8. After most of the sample has passed through the filter add 1 - 2 ml saturated $MgCO_3$ solution.
9. Remove the filter, using tweezers - avoid touching it with your fingers - they may have acids on them.

Filter Handling - Distant Samples

1. Fold each filter in quarters and place in aluminum foil and label with date, location, sample number, volume of sample filtered and "Phyto".
2. Freeze filters and ship to lab regularly on dry ice.

Filter Handling - Local Samples

1. Fold each filter and place it in a numbered culture tube.
2. Record the volume of sample, sample number, date and the tube number on the chlorophyll-a data sheet.
3. Place each tube sequentially into a tube rack and immediately place in the freezer in the Phosphorus lab. Put chlorophyll data sheet into the in basket.

2.8 Water Sampling Under Ice

General Considerations

1. Sampling locations to be sampled in the winter should be located as close as possible to the open-water stations.
2. Clear loose ice and snow; drill through the ice with a hand or motorized auger. Special effort must be made to ensure auger shaft is kept clean. Keep the area around the hole clear of potential contamination (e.g., dirt, fuel, oil, etc.). Avoid gas, oil and exhaust contamination of the sampling equipment.
3. Remove all ice chips and slush from the hole using a plastic sieve.
4. Samples should be collected approximately 0.2 m below the bottom of the lake ice using a depth sampler (e.g., Van Dorn sampler) or a GeoPump® sampler to minimize the possibility of contaminant introduction associated with augering if a motorized auger is used. Don't let the sampler come into contact or stir up sediment from the lake bottom as this will contaminate the water sample.
5. In extreme cold temperatures (well below freezing), do not rinse the sample bottles, the rinse water will freeze to the surface of the bottle. Send a bottle blank in for analysis when these conditions exist.
6. Ice thickness and total depth should be measured.
7. Otherwise follow the sampling procedures outlined for open water sampling using the GeoPump® and depth samplers.

All sample bottles should be clearly labelled with date, location, site, depth, analytical parameter group and sampler ID. Store and transport all samples at 4 °C in a closed cooler. **Do not allow the samples to freeze.**

Reference

This protocol was derived from Environment Canada (1983), BC-RISC (1997a), RAMP (2005), and EMAN-North (2005).

2.9 Zooplankton Sampling Methods

Purpose

Zooplankton are small invertebrates that float freely in the water column of lakes and oceans. Zooplankton in lakes range from a few tens of microns (Protozoa) to >2 mm (macrozooplankton). The dominant taxonomic groups are Crustacea and Rotifera and so sampling protocols tend to quantify those groups, typically using zooplankton net sampling techniques. Zooplankton are important as both prey and consumers in the aquatic food web, and as biomonitors because they are also highly sensitive to environmental change or disturbance in lakes (Paterson 2003). Zooplankton are sampled to provide quantitative estimates of community composition, densities and/or biomass within lakes. Zooplankton densities and species composition show spatial variability both horizontally and vertically in lakes.

General

- CAUTION: FORMALIN IS A SUSPECTED CARCINOGEN - USE IT WITH CARE AND READ MSDS.
- This technique is normally used at the deepest site of the lake to capture the vertical distribution of zooplankton, if only one site is to be sampled. It should be noted that species typical of deeper waters may be over-emphasized.
- Check for holes or tears in the net before the field trip.
- Rinse the net with tap water and hang to dry at the end of the day.
- Duplicate hauls may be required – depending on the study design (check with project manager).
- Supporting information at each site may include *in situ* water quality measurements, chlorophyll-a and phytoplankton species composition estimates.
- Zooplankton typically migrate vertically during the day and night. Thus, it may be beneficial in terms of consistency, to sample those sites intended to be sampled multiple times, at approximately the same time of day.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Zooplankton net (63 µm mesh), bucket and line marked for depth. The filtering area of the net should greatly exceed the area at the mouth of the net to minimize clogging and loss of the sample.
- Buffered Formalin (see Appendix C) and glycerin
- Labelled jars - Wheaton snap cap 150 ml
- Wash bottle
- Labelling tape and waterproof markers and field sheet/book
- GPS unit, depth sounder, camera, tape measure.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals

Procedure

1. Soak the body of the zooplankton net in lake water prior to use (2 min).
2. Rinse the net with lake water to dislodge any attached material prior to sampling.
3. Attach the zooplankton bucket - make sure the plug is in place.
4. Fill the Nalgene® squirt bottle with lake water that has been filtered through the net mesh.
5. Lower the net to the euphotic zone depth making sure it stays in a vertical position.
6. Raise the net vertically at a continuous rate of 0.5 m/sec to minimize avoidance of the net by fast-swimming zooplankton.
7. Avoid sampling near sediments and macrophytes because non-planktonic species of Rotifera and Crustacea inhabit these substrates and would contaminate the planktonic sample.
8. At the surface, rinse down the outer sides of the net two to three times with lake water. Do not splash rinse water into the net opening, or let it drop below the surface.
9. Separate the bucket from the net, place the lower end of the bucket into an open sample jar, then remove the plug and drain the zooplankton and water into the jar.
10. Rinse the bucket contents into the sample jar with squeeze bottle previously filled with filtered net water.
11. Zooplankton samples can be preserved in either 95% ethanol or 5% formaldehyde. Formaldehyde is preferred because counting samples preserved in ethanol is difficult because of the convection currents caused by rapid evaporative losses. The following approaches can be used to reduce distortion due to Formalin preservation: (a) addition of 40 g/L of sucrose to formaldehyde solutions; (b) maintenance of samples at low temperature (6°C); and (c) narcotisation with carbonated water or methanol prior to preservation in a formaldehyde-sugar solution (Paterson 2003).
12. Rinse the net and bucket with lake water between sites.
13. Record sampling location (GPS coordinates), site, date, time, samplers ID, number of hauls and depth of haul on the jar and field sheets/book. Also note the mesh size and dimensions, the fixative used and the prevailing weather conditions in the field sheets/book. Note and record ice depth in the winter.
14. Put a few drops of glycerin into sample when back at lab, before storage of sample. This helps prevent the animals from sticking together.

Composite Zooplankton Sampling

This technique has been instituted to account for more of the spatial heterogeneity of a typical lake; one net haul at the main site (as per the previously described method) might not be an accurate reflection of this. This technique is merely an extension of the previous method, utilizing the same equipment and sampling technique. Some additional notes to keep in mind are elaborated on below.

1. Compositing the samples from 10 (or 15 sites if Secchi warrants it) sites will have to be done into a 1 litre bottle (plastic, preferably). Ensure boat is anchored at each site unless wind conditions are calm enough to allow the net to be lowered and raised straight down and up.
2. Ensure that depth of haul is noted on field sheet from each of the 10 haul sites to enable accurate calculations of community composition.

3. Ensure that after first haul, that buffered Formalin is added to the 1 litre container, and continue to top this up as you sample the other haul sites such that the total preservation concentration is 7 to 10 percent of total plankton-water volume.
4. When back at the lab, use a paster (sieve constructed of PVC and 63 µm Nitex® mesh) to gently concentrate the zooplankters into the usual 150 ml Wheaton jar. Do this in a fume hood with appropriate PPE. Preserve this final sample with 7 – 10 percent buffered Formalin and add a few drops of glycerin.
5. Enter the filtered volume on the Wheaton jar, along with other pertinent information.

Reference

This protocol was derived from Alberta Environment (2002) and Paterson (2003).

2.10 Bacteriological Sampling

Purpose

Bacteriological samples are taken to assess the microbiological and sanitary quality of the water, and to assess the potential health risk from waterborne diseases. Samples are typically analysed for a combination of the following bacterial parameters: total (rarely) and faecal coliforms, *Escherichia coli* (*E. coli*), fecal streptococci and enterococci. Due to the high risk of potential contamination of the sample during collection, care must be taken when collecting bacteriological samples and additional procedures are followed to try and maintain sterile conditions.

General

- When sampling from a boat, obtain the sample from the upstream side of the boat.
- Keep the sample at 4 °C and deliver it to the Provincial Laboratory of Public Health within 24 hours. Do not freeze.
- Affix the request sheet number to the bottle upon return to the office.
- Complete the Request for Bacteriological Examination of Water form ensuring that the form and bottle number are identical. Ensure that the sample number, site name, date, and time are clearly marked on the sheet. Time is recorded in MST. The request sheet must be marked with "analyze to endpoints".
- Samples are not accepted by the lab after noon on Friday, or weekends, unless special arrangements are made as media must be prepared (especially for *E. coli*).
- When collecting more than a few samples, contact the Provincial Lab prior to the sampling trip and let them know the sample number and type to be collected.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Sterile bacteriological bottles supplied by the analytical laboratory.
- Sample request sheets and Chain of Custody forms (COCs)
- Depth sampler (e.g., Van Dorn, Kemmerer) - for discrete deep water sites or tube for composite sampling.
- Labelling tape and waterproof markers and field sheet/book
- GPS unit, depth sounder, camera, tape measure.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).

Procedure

Shore Sampling

1. Wade out to knee deep water beyond the point where wave action affects the lake bottom (avoid contamination by suspended sediments).
2. Do not disturb the sediment/substrate. Wait 2-3 min to ensure any sediment disturbed by wading has settled.

3. Do not rinse the bottle or touch the inside of the bottle or cap. Always hold bottle upright and by the base, as the preservative is already in bottle. Keep sample bottle closed until needed.
4. Holding the bottle upright and by the base, submerge till the bottle opening is approx. 30 cm below the water surface, facing towards the current. Uncap and fill the sample bottle to the 200 ml line, cap and bring to the surface. If the bottle is filled slightly above the 200 ml line carefully pour the excess out and cap. If the bottle is completely full, resample with another bottle. Immediately place the bottle in a closed cooler with ice packs or hot water bottles, depending on the season.
5. If necessary, the sample bottle can be filled from a clean (sterile) intermediate container.
6. Take several individual samples along the length of the beach.
7. To take a composite integrated sample: fill one sample bottle to 200 ml at each haul site and average the results, instead of collecting a partial bottle at each site. If necessary, the sample bottle can be filled from the composite sample bucket.

Offshore Surface Sampling

1. Sample from the bow of the boat to prevent potential contamination from the boat or the motor.
2. Take a sample at arms length from the boat following steps 3 and 4 from the shore sampling protocol. Sample towards the current (the direction the boat is facing).

Offshore Sampling at Depth

1. Collect a sample of water at the desired depth with a depth sampler according to Section 2.4.
2. Do not rinse the bottle or touch the inside of the bottle or cap, and always hold bottle upright and by the base, as preservative is already in bottle. Keep sample bottle closed until needed.
3. Fill the sample bottle to the 200 ml line and immediately cap the bottle securely. Immediately place the bottle in a closed cooler with ice packs or hot water bottles, depending on the season.

Reference

This protocol was derived from BC-RISC (1997b), Alberta Environment (2002), USGS (2005), Calgary Health Region (2005) and EMAN-North (2005).

2.11 Sediment Coring

Purpose

Core samplers are tube-like devices that penetrate the sediment by gravity (free-fall), vibration, hydraulic pressure (water or oil) or by hand (scuba divers). Sediment cores are taken to determine recent and/or historical sediment physicochemical conditions in depositional aquatic environments. A sediment core provides a vertical sediment profile with the period of sediment deposition correlated with core depth unless significant mixing has occurred. Cores should be taken where stratification of the sediment is suspected or known and/or historical sediment quality is of interest.

Cores are sectioned, and the upper 2-10 cm (depending on design details) of the sediment core and/or various sections from the lower part of the core submitted for physicochemical analysis. Core sections can also be submitted for geological characterizations and radioisotope dating (e.g., Pb210 dating) to determine the period of sediment deposition. The latter provides a measure of the rates of sediment deposition over time as well as estimates of the age of various sediment layers. This information can then be combined with chemical measurements to provide estimates of how sediment quality may have changed over time.

There are several types of sediment corers and selection depends on the nature of the water body sampled, the study objectives, reliability and logistics. Corers are efficient at sampling all sediments except for sand, gravel, firm clay or till, where grab samplers may be preferred depending on study objectives. However, some core samplers such as the vibra-corer, are better suited to coarse or compacted sand compared to other corers.

General

- Samples should be taken from sites, progressing in order, from the least contaminated site first to the most contaminated site last. The degree of contamination can be estimated from historical data, site conditions, land use, professional knowledge etc.
- Repeat the coring procedure if insufficient sample is collected on the first try. Check how much sample is required by the analytical laboratory ahead of time.
- Do not core the same spot over again. Use both sides of the boat and/or drift back on the anchor line before repeating core.
- To protect hands, wear gloves when raising and lowering the corer.
- Allow sufficient room in the sample containers for expansion upon freezing.
- The type of sample container and slicer used will depend on laboratory requirements. Cores may be collected for nutrients, trace organics, trace metals etc.
- In shallow water, cores can be collected with a core tube hose clamped to the bottom of an extendable paint pole, with the top of the tube sealed onto a one-way valve.
- See Section 4.3 for sampling equipment cleaning and decontamination procedures.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Sediment core sampler with messenger (Kajak-Brinkhurst Corer [KB] or the 4-bore corer [4-bore]).
- Core liners with bottom O-ring and end caps. The type of liner depends on the sediment parameters to be measured (e.g., cellulose acetate butyrate [CAB] stainless steel, polybutylacrylic, Teflon®). Use stainless steel for trace organic sampling, plastic liners for trace metal sampling.
- Core extruder and slicer. The type of core extruder and slicer depends on the sediment parameters to be measured (e.g., Teflon®, polyethelyne, stainless steel). Use stainless steel for trace organic sampling and plastic liners for trace metal sampling.
- Labelling tape, waterproof pens and fieldbooks/sheets.
- Hydrographic map showing sample sites.
- Leak-proof sediment sample containers (e.g., widemouth glass jars, Teflon® lined lids).
- Coolers with ice packs or hot water bottles, depending on the season.
- Ziploc® bags, a pack of disposable syringes or turkey basters.
- Lab analysis request sheets and Chain of Custody forms (COCs).
- GPS unit, depth sounder, ruler, tape measure, camera.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Equipment for field cleaning/decontamination procedures (see Section 4.3)
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals

Procedure

This procedure is specific to the two types of gravity corer: the K-B corer or the 4-bore corer. Gravity corers are released at the water surface, allowed to free-fall and penetrate the sediment under the samplers own weight. By contrast, other types of corer such as vibro-corers are vibrated into the sediment. Sampling procedures for several types of sediment corers are given in USEPA (2002) and Ohio EPA (2001). A typical gravity core sampler is a length of cylindrical pipe with a weighted head. A liner which is usually made of plastic is inserted to hold the sample. The type of liner appropriate for sampling depends on the sediment variables to be analysed. At the bottom end of the sampler is a metal core cutter which facilitates sediment penetration (nose piece), and a core catcher to retain the sediment in the liner. At the top end of the sampler is a ball-valve or piston which retains sediment in the liner when the sampler is retrieved from the sediment.

Core Collection

1. Pre-label the sediment containers with site, date, location and depth of slice.
2. Place a clean liner in the corer or each of the four barrels of the corer, depending on the type of corer being used.
3. Push the liner in until the 0-ring fits snugly - the liner should protrude 2-5 cm at the bottom.
4. Set the stoppers in the open position.

5. Lower the corer over the side of the boat (ensure that the rope is securely attached to the corer). Lower the corer slowly to minimize the creation of shock waves at the front of the sampler that may disturb and re-suspend fine sediments.
6. Let the corer slowly sink straight into the sediment.
7. Release the messenger.
8. Raise the corer after the messenger releases the stoppers.
9. Just before the bottom of the corer breaks the water surface, have a second person reach under and quickly cap the cores with the insert liners.
10. Raise the unit into the boat, keeping it in the upright position.
11. Remove each liner from the bottom of the corer - be careful not to spill any of the enclosed water.
12. Cap the top of each core tube and place in holder.
13. Taking one core at a time, remove the bottom cap and quickly replace it with the core extruder. Keep constant pressure on the top cap when doing this to help form a vacuum.
14. Core samples will be deemed acceptable if the core was inserted vertically into the sediment, adequate depth was sampled and there was no sediment loss.

Recording of Core Characteristics and Supporting Information

Photographs, field notes and measurements should be taken and logged during sediment core collection. These will include, but may not be limited to:

- **Sediment Core Photographs**: the core should be photographed twice with a digital camera; once in ambient light and once using a fill-in flash. The core sample should occupy a minimum of 70% of the image and a label and a scale (ruler) should be included in the view. Additional photographs should be taken if any anomalies or artifacts are encountered.
- **Sediment Core Measurements/Observations**: total core depth; vertical profile/structure (i.e., depth and description of distinct layers); type of material (soil type, colour, moisture condition, density, and grain size), biological structure (e.g. shells, large tubes, biota, macrophytes); debris (e.g., wood chips, plant or other fibres; obvious signs of anoxia (e.g., black layers); degree of sample disturbance; obvious odour or oily sheen; other unusual properties.
- **Sampling Site Description**: target and actual sampling location (GPS); date and time of sample collection; overlying water depth (m); ambient weather conditions; core penetration depth, sampling personnel; any deviations from the field sampling procedure.

Core Sectioning

1. Remove the top cap from the tube and siphon off excess water. Carefully push the sediment core to the top of the core tube, expelling any remaining excess water.
2. Set the core slicer on the top of the sediment tube.
3. Push the sediment into the core slicer and cut off the required amount of sample -usually the upper 4-6 cm but can be between 2-10 cm of the upper layer.
4. Place the sample in a labelled sample container. Place each sample container in two Ziploc® plastic bags (double-bag) in case of leakage.
5. Repeat the extruder steps for each of the four tubes (if using the 4 barrel corer).

6. Rinse the tubes and corer with lake water before collecting new samples or decontaminate the equipment between sites (see Section 4.3). The project manager should decide which procedure is applicable to the sampling project.
7. Store and transport samples in a closed cooler samples at 4 °C and do not allow to freeze unless intended to be stored frozen.
8. If samples are not to be frozen and are to be stored at 4 °C until subsequent analysis, the sample containers should be filled to the rim with no headspace, to reduce oxygen exposure. This is particularly important if volatile parameters are to be measured.
9. If samples are to be frozen then a headspace of approximately 10% of the jar volume should be left in glass containers, to accommodate expansion of the sample when frozen. Check with the analytical laboratory before sampling to confirm sample storage requirements for the analyses requested. Samples stored at 4 °C in the dark can generally only be stored for days or weeks before analysis except for particle size analysis (grain size) which can be stored for up to 6 months. Samples can be frozen and archived for longer periods of time but it should be noted that samples destined for particle size analysis should not be frozen. See Environment Canada (2004) and USEPA (2002) for further information regarding sample storage and archiving.

Core Compositing

If the sediment collection from a particular horizon (e.g., the upper 2-5 cm) is of insufficient volume to fulfil analytical requirements then several cores should be taken at the same site. The relevant core horizons should be sectioned and combined and then sub-sampled as soon as possible. It is recommended that only horizons of the same depth and similar stratigraphy be composited (see USEPA 2001 for further details).

1. Combine the similar sediment horizons from different cores in a stainless steel bowl. Remove any debris and/or biological materials if request by project manager.
2. Once sufficient sediment is collected, stir with a stainless steel utensil the composite sample for 30 sec, and then transfer into the appropriate pre-labelled containers with a stainless steel or Teflon® implement. Samples should appear homogenous (i.e., uniform colour and texture). Avoid over-mixing.
3. Store and transport samples in a closed cooler at 4 °C and do not allow to freeze unless intended to be stored frozen. Place each sample container in two Ziploc® plastic bags (double-bag) in case of leakage.
4. If samples are not to be frozen and to be stored at 4 °C until subsequent analysis, the sample containers should be filled to the rim with no headspace, to reduce oxygen exposure. This is particularly important if volatile parameters are to be measured.
5. If samples are to be frozen then a headspace of approximately 10% of the jar volume should be left in glass containers to accommodate expansion of the sample when frozen. Check with the analytical laboratory before sampling to confirm sample storage requirements for the analyses requested. Samples stored at 4 °C in the dark can generally only be stored for days or weeks before analysis except for particle size analysis (grain size) which can be stored for up to 6 months. Samples can be frozen and archived for longer periods of time but it should be noted that samples destined for particle size analysis should not be frozen. See Environment Canada (2004) and USEPA (2002) for further information regarding sample storage and archiving.

Reference

This protocol was derived from Ontario MOE (1996), Ohio EPA (2001), Alberta Environment (2002) and USEPA (2002).

2.12 Sediment Grab Sampling

Purpose

Sediment dredging or grab sampling techniques are used to collect surficial sediment samples. This type of sampling is conducted when the quality of recently deposited sediments is of interest, historical sediment quality is not of interest, and relatively large volumes of sediment are required. Grab samples can be easily taken in depositional shallow sediments or in relatively coarse sediments, where core samples cannot be obtained.

General

- This procedure is used if there is insufficient sediment to use a core sediment sampler and for a variety of other reasons (e.g., the need to define sediment quality as it relates to benthic invertebrates).
- Samples should be taken from sites, progressing in order, from the least contaminated site first to the most contaminated site last. The degree of contamination can be estimated from historical data, site conditions, land use, professional knowledge etc.
- At any particular site, water sampling should be conducted prior to sediment sampling to avoid disturbance of overlying waters by the sediment sampling technique.
- If the jaws are not closed properly when the dredge is retrieved, discard the sample and re-sample.
- See Section 4.3 for sampling equipment cleaning and decontamination procedures.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Dredge sampler - Ponar or Ekman grab sampler.
- Rope (non-twisting nylon).
- Stainless steel or glass bowl.
- Stainless steel or Teflon®/plastic spoons and slicers.
- Labelling tape, waterproof pen and note pad.
- Hydrographic map showing sample sites.
- Leak-proof sediment sample containers (e.g., widemouth glass jars, Teflon® lined lids; material of containers would depend on analysis).
- Coolers with ice packs or hot water bottles, depending on the season.
- Ziploc® bags, a pack of disposable syringes or turkey basters.
- Lab analysis request sheets and chain of custody forms (COCs).
- GPS unit, depth sounder, tape measure, camera.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Equipment for field cleaning/decontamination procedures (see Section 4.3).
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals.

Procedure

1. Label the sample containers with site identification, sample type, sampling method, sampler ID and the date of collection.

2. Note the following site/sampling information in the field sheet/book during the sampling process: target and actual sampling location (GPS); date and time of sample collection; overlying water depth (m); weather conditions; sampling personnel; any deviations from the field sampling procedure, macrophyte growth.
3. Ensure that the dredge jaws open and close properly.
4. Lock the dredge jaws in the open position and lower in a controlled fashion to the lake bottom. Do not allow the sampler to "free fall" The sampler should be in contact with the substrate or positioned just above it.
5. Drop the messenger (if applicable) and slowly raise the sampler off the bottom to prevent loss of fine sediment and then raise the dredge to the water surface.
6. The sample is deemed acceptable if the desired depth of penetration has been achieved; and the sampler has completely closed and was not inserted on an angle or tilted upon retrieval. If the sample does not meet these criteria the sample should be taken again close to the original sampling location. The rejected sample should be discarded in such a way that it will not affect subsequent sampling efforts.
7. The actual achievable penetration depth depends on the nature of the sediment and the sampling device used. A minimum penetration depth of 6-8 cm is recommended for surficial sediment samples but the preferred depth is 10-15 cm. These depths ensure minimal disturbance to the upper 2-5 cm of sediment that will be removed from the grab sample and submitted for physicochemical analysis.
8. Note the following sediment measurements/observations (where applicable): grab penetration depth; depth sub-sampled; type of material (sediment type, colour, moisture condition, density, and grain size), biological structure (e.g. shells, large tubes, biota, macrophytes); debris (e.g., wood chips, plant or other fibres; obvious signs of anoxia (e.g., black layers); degree of sample disturbance; obvious odour or oily sheen; other unusual properties.
9. Siphon off any water on the surface of the grab sample with a syringe but if the water is cloudy allow it to settle first (use a new syringe for every site). Remove the upper 2-5 cm of sediment with a stainless steel or Teflon® implement and transfer to a stainless steel/plastic tray/bowl. Avoid sediment at the edges of the grab sample (touching the grab sampler).
10. If more sediment is required to obtain the required volume for analysis, then more grab samples should be collected from the same site in undisturbed sediment. The volume of sediment to be submitted to the analytical laboratory depends on the study objectives, the parameters to be analysed and the analytical laboratory. The composite sample tray/bowl should be covered while grab samples are being collected. The number of grab samples collected should be noted.
11. Wash the dredge off in the lake. Rinse bucket and ladles before and after each site with lake water. If sampling for organics, do a hexane/acetone rinse, collecting the waste in a jar for transport back to lab. Aluminum foil used to cover the bucket must also be rinsed with hexane/acetone (see Section 4.3 for detailed decontamination procedures).
12. Once sufficient sediment is collected, stir (homogenize) the composite sample for 30 sec, then transfer into the appropriate pre-labelled containers with a stainless steel or Teflon® implement.
13. Store and transport samples in a closed cooler at 4 °C and do not allow to freeze unless intended to be stored frozen. Place each sample container in two Ziploc® plastic bags (double-bag) in case of leakage.

14. If samples are not to be frozen and to be stored at 4 °C until subsequent analysis, the sample containers should be filled to the rim with no headspace, to reduce oxygen exposure. This is particularly important if volatile parameters are to be measured.
15. If samples are to be frozen then a headspace of approximately 10% of the jar volume should be left in glass containers, to accommodate expansion of the sample when frozen. Check with the analytical laboratory before sampling to confirm sample storage requirements for the analyses requested. Samples stored at 4 °C in the dark can generally only be stored for days or weeks before analysis except for particle size analysis (grain size) which can be stored for up to 6 months. Samples can be frozen and archived for longer periods of time but it should be noted that samples destined for particle size analysis should not be frozen. See Environment Canada (2004) and USEPA (2002) for further information regarding sample storage and archiving.

Reference

This protocol was derived from Ontario MOE (1996), Ohio EPA (2001), Alberta Environment (2002), USEPA (2002), Environment Canada (2004) and RAMP (2005).

2.13 Macrophyte Sampling Methods

Aquatic macrophytes have historically received less attention than other aquatic components, and standardization of field sampling protocols is generally lacking. Standardized sampling protocols do not currently exist within any Alberta or federal agency. Other jurisdictions, particularly in the United States, have developed regional protocols, but these vary widely in approach and level of effort. The methods described in this section represent an amalgam of elements from various approaches, selected on the basis of applicability to Alberta conditions.

Purpose

Aquatic macrophytes are sampled for a variety of purposes, including:

- species inventories (presence/absence);
- identifying invasive species;
- biodiversity studies;
- aquatic health assessments;
- primary productivity assessments; and
- biomonitoring to determine the effects environmental change or anthropogenic stressors.

Field studies may be qualitative or quantitative in nature and appropriate survey methodology is typically selected on the basis of project-specific objectives.

General

The aquatic macrophyte sampling protocols described in this document are applicable to surveys conducted on wetlands, ponds, lakes and reservoirs. To a lesser extent, they may also apply to large river surveys, discussed in Section 3.8. Although the selection of specific survey method(s) will depend largely on project objectives (Table 2.13-1), consideration must also be given to the type of water body (i.e., lotic vs. lentic, size of water body or study area; littoral extent; and depth) and the nature of the aquatic plant communities to be sampled.

Table 2.13-1 Summary of recommended aquatic macrophyte survey methods in relation to survey objectives

Objective	Methods	Description
Reconnaissance	Surface Inventory	Qualitative
Species Inventory/Biodiversity	Surface Inventory	Qualitative
	Point Intercept	Qualitative/Semi-Quantitative
	Line Intercept	Qualitative/Semi-Quantitative
Biomass/Productivity/Biomonitoring	Transect with Quadrat	Quantitative

The biomass and species composition of lentic aquatic macrophyte communities are influenced by a wide variety of factors, including: water quality and clarity; substrate; nutrients; temperature; wave exposure; slope; depth; and pressure. Water bodies with diverse habitat types will generally support more diverse aquatic macrophyte communities than water bodies with relatively homogenous habitat types. Diversity of community types and sampling conditions may, in turn, influence the selection of the appropriate sampling method and study design (e.g., survey density or number of samples taken).

Field staff should be familiar with regional aquatic macrophyte species and the use of plant identification keys. Some examples of useful keys for Alberta are provided in the Reference Section at the end of this protocol. The sampling methods discussed in this document are intended for sampling in open-water habitats, where plants fall into three generalized categories:

- *submergent* - entire plants submerged;
- *floating-leaved* - plants with submerged parts and leaves that float at the surface; and
- *emergent* – plants with erect parts (stems and leaves) that rise above the water surface.

Emergent species or communities, and the first two categories to a lesser extent, may form a transition between open water and riparian or marsh habitats which are not covered by the methods discussed here. Within all three categories, all rooted vascular plants should be considered. Macroalgae (i.e., *Chara* sp. and *Nitella* sp.) are similar to vascular aquatic macrophytes in size, form and function and should be treated the same. Filamentous algae, aquatic mosses and free-floating vascular plants (e.g., *Lemna* sp.) should be recorded when present, but quantification is subject to specific study design.

Plants should be identified in the field whenever possible, but field staff should also be knowledgeable in the preparation and preservation of aquatic macrophytes (Haynes 1984; Warrington 1994; Parsons 2001) for office identification or submission to a qualified plant taxonomist. Specific study designs may also require retention of reference collections. Field staff should be aware of and able to identify invasive/exotic species, as well as rare or endangered species. The Alberta Natural Heritage Information Centre (ANHIC) maintains tracking and watch lists for rare Alberta species. These list, as well as reporting procedures and report forms, may be obtained at http://www.cd.gov.ab.ca/preserving/parks/anhic/plant_trk_wtch.asp.

For many studies it may be adequate to collect information that provides comprehensive species lists only, or delineates bed types and bed distribution. Many studies, however, require a correlation between plant presence/absence or plant bed type and other habitat parameters. Water depth, Secchi depth, light regime and water temperature are measurable parameters that are commonly associated with lentic aquatic macrophyte surveys. Another key parameter is substrate type, as it is often a key factor determining plant distribution. Substrate type can be described in terms of hardness, embeddedness (the degree to which fine sediments surround coarse substrates), organic content and substrate composition. Description of substrate composition is provided in Section 3.8. Basic field description of substrate type can be based on prodding with a pole or paddle, but a more detailed description requires the collection of a sample with an Ekman dredge or comparable bottom grab sampler (Section 2.12).

Timing of aquatic macrophyte surveys may be dependent on study objectives and design, but in most cases it is appropriate to conduct surveys during the peak growing season (i.e., mid-summer to early fall). Surface inventories or other reconnaissance surveys may be conducted outside of the peak growing season, provided that plant growth has progressed enough to permit determination of bed extent and species identification. However, it is important to note that many aquatic macrophytes are difficult to identify prior to development of inflorescences or seed.

Equipment

Surface Inventory

- study area maps (bathymetric maps if available)
- personal protective equipment (PPE) appropriate to the conditions (e.g., personal floatation device[PFD])
- boat, capable of manoeuvring in shallow water (with outboard motor or jet drive where appropriate and practical)
- double-headed rake (two garden rake heads welded back to back), with long or telescopic handle or throw rope, as appropriate for depths to be encountered
- clear, sealable plastic bags (medium or large size)
- waterproof labels for voucher samples
- waterproof markers, pencils, water resistant field note books or field sheets with clipboard
- Secchi disk
- light meter or turbidity meter (optional)
- Ekman dredge, or similar grab sampler (optional)
- sounding line, depth sounder or depth gun
- underwater viewer (optional)
- polarized sunglasses
- thermometer
- GPS unit (with tracking capability preferred)
- digital camera
- hand lens or field microscope (optional)
- plant identification keys (optional); and
- plant press (optional)

Point Intercept

Equipment requirements are generally the same as for a Surface Inventory, with the following exception:

- GPS unit with horizontal accuracy of 5 m or better (real-time navigation capability preferred)

Line Intercept

Equipment requirements are generally the same as for a Surface Inventory, with the following exception:

- transect line, marked at 1 m intervals with fluorescent flagging tape. The transect line should be of a set length (e.g., 100 m), appropriate for the size of the water body or plant beds likely to be encountered. Alternatively, line length may equal the distance between end points in studies with permanent transects.

Transect with Quadrat

Equipment requirements are generally the same as for a Surface Inventory, with the following exceptions:

- chest waders, for shallow waters that can be waded safely
- diving equipment for two divers, according to safe diving procedures and Alberta Environment policy (e.g., PADI)
- sampling quadrat (e.g., 1 m x 1 m sq.)
- scissors or garden shears capable of cutting coarse vegetation
- large fine-mesh bag
- centrifuge dryer
- portable, water resistant, battery-powered balance(s) (capacity and precision dependent on volumes of plants anticipated and specific study designs)

Procedure

Surface Inventory

Surface inventories provide a qualitative method of collecting data for production of plant species or community distribution maps. In many cases, surface inventories may serve as reconnaissance surveys that are adequate to document gross changes in community structure or extent of plant beds over time. Further, surface inventories can serve as reconnaissance surveys conducted as an initial step in support of more intensive aquatic macrophyte surveys.

1. Perform a preliminary determination of the littoral zone (the shallow, usually nearshore, regions of a water body, where light penetrates to the bottom permitting colonization by rooted aquatic macrophytes and benthic algae). Small or shallow water bodies may be littoral throughout, while large or deeper water bodies may be littoral only around the margins or in shoal areas. Maximum depth of aquatic macrophyte growth is usually

limited by light penetration (i.e., the euphotic zone), but may also be limited by other factors such as slope and substrate type. In rivers, aquatic macrophyte growth is often limited by substrate type and flow velocity, and plant beds are usually restricted to river margins.

2. Littoral regions around lake shores or along river banks are surveyed by navigating the boat in a zig-zag pattern; from shallow nearshore waters out to the extent of plant beds (Figure 2.13-1). Pattern density (i.e., distance between passes) is dependent on visibility, but should allow complete coverage for visual assessment. Shoal areas with aquatic plant growth should be surveyed separately. Shorelines without plant growth need not be surveyed.
3. Shallow water bodies with littoral plant growth throughout can be surveyed in a grid pattern, with grid density dependent on study design, area to be surveyed or time constraints.
4. GPS tracking feature (if available) should be employed to provide an accurate record of survey pattern and area traversed. All key features, such as plant bed boundaries or transitions from one plant community type to another, should be recorded as UTM GPS waypoints and described in a notebook or on field sheets.
5. Water depth, Secchi depth, and turbidity and bottom light level (if equipment is available) should be recorded at all key feature waypoints. These parameters, along with GPS coordinates, should also be recorded periodically at representative sites within plant beds. Water temperature or temperature profiles should be recorded at several shallow and deep sites.
6. To the extent possible, plants occurring within beds should be identified to species and recorded. For a qualitative inventory survey it is adequate to record presence only.
7. In shallow water with adequate visibility, plants may be identified by observation from the boat. In deeper or murkier water, or where a canopy of plants obscures plants at lower levels, an underwater viewer is a useful aid.
8. At intervals within each plant bed, plant samples should be collected with the rake sampler for closer examination. Plants may be identified on site or they may be retained for later identity verification. Collected plants should be placed in a sealable plastic bag, along with a label providing all pertinent information. Sample collections should also be recorded in a note book or on field sheets, along with a sample number, all pertinent site information and GPS location.
9. Additional information from very shallow nearshore waters may be obtained from shore or by wading.
10. Samples to be archived or included in reference collections should be properly preserved in a plant press.

The end products for a surface inventory include a map showing the distribution of plant bed types, and a list of species for each plant bed type and the water body or study area as a whole.

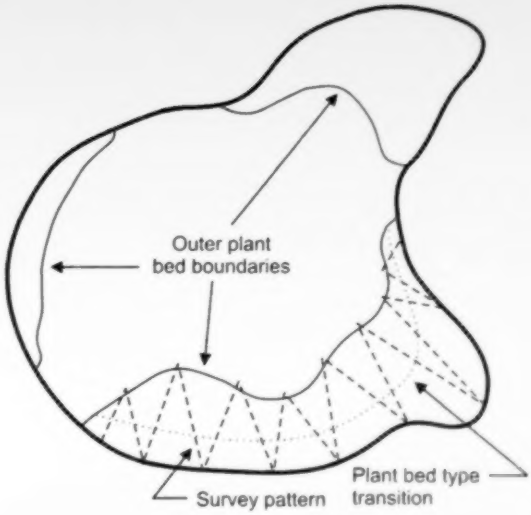

Figure 2.13-1 **Example of a possible survey pattern for a surface inventory conducted on a small lake**

Point Intercept

Point Intercept surveys are conducted by sampling for aquatic macrophytes at regularly spaced, pre-selected points in a grid pattern. Surveys may be conducted over an entire water body or a discrete area within a water body, but either way the objective is to survey an entire study area without reliance on subjective site selection in the field. Grid node (i.e., sampling site) coordinates can be determined manually from maps, or generated with the aid of GPS or GIS software packages. Sampling sites are then entered as UTM waypoints into a GPS unit or system, preferably one with real-time differentially corrected navigation capability.

1. Navigate to each pre-selected point in a regular pattern. Sampling sites located in shallows states may be sampled by wading.
2. Water depth, Secchi depth, and turbidity and bottom light level (if equipment is available) should be recorded at all sampling sites. Water temperature or temperature profiles should be recorded at several shallow and deep sites within the study area.
3. Record the species present at each sampling site based on observation from the boat. Additional information is acquired using an underwater viewer and rake sampling.
4. Plants may be identified on site or they may be retained for later identification. Collected plants should be placed in sealable plastic bags, along with a label providing all pertinent information. Sample collections should also be recorded in a note book or on field sheets, along with a sample number, all pertinent site information and GPS location.
5. Samples to be archived or included in reference collections should be properly preserved in a plant press.

The survey data can be used to identify and delineate plant communities or bed types. The end products for a qualitative Point Intercept survey include a map showing the distribution of plant bed types, and a list of species for each plant bed type and the water body or study area as a whole.

The preceding methodology describes a qualitative survey. Semi-quantitative results can be obtained through modification of the study design and the field sampling procedure as follows.

1. Conduct sampling at each sampling site in a consistent manner and with a consistent level of effort. At each site, the same number of rake drags or rake tosses should be made and, to the extent possible, each drag or toss should sample an equivalent area.
2. At each sampling site, determine and record the relative robustness of plant growth. Example descriptors could include Dense, Moderate, Sparse and Trace.
3. For each rake sample, identify the species present and estimate the relative proportion of each species in the sample. Species that are present in very small amounts, perhaps only as fragments, can be recorded as 'present' or 'trace' only.

Line Intercept

The Line Intercept method utilizes a system of transects that representatively encompass all aquatic macrophyte community types within a water body to derive a qualitative description of those communities. Transects are generally laid out perpendicular to the shore.

1. Conduct a preliminary reconnaissance survey (e.g., Surface Inventory) to identify and delineate all vegetated littoral areas within the water body or study area. The number of transects required will vary from site to site, and according to study design. Stratified sampling designs are generally most appropriate.
2. Using a braided nylon rope, prepare a transect line marked at 1 m intervals with fluorescent flagging tape. Use of alternate colours at the 5 m and 10 m intervals simplifies tracking along the transect line. The transect line should be of a set length (e.g., 100 m), appropriate for the size of the water body or plant beds likely to be encountered. Alternatively, line length may equal the distance between end points in studies with permanent transects.
3. Secure the transect line at both ends by attaching to poles or to anchor lines.
4. Surveys are conducted by navigating the boat along a transect line and recording all species occurring along individual line segments. Plants are considered present if they intersect the vertical plane between the transect line and the bottom.
5. In some cases wading is an option in very shallow nearshore waters. In deeper or murkier water, or where a canopy of plants obscures plants at lower levels, an underwater viewer is a useful aid. Rake sampling may also be used to collect samples for observation or reference collection.
6. Water depth should be recorded at each end of the transect line and at interval markers. GPS waypoints, Secchi depth, and turbidity and bottom light level (if equipment is available) should be recorded at each end of the transect line, and may also be recorded at intervals along the line (e.g., at important transitions). Water temperature or temperature profiles should be recorded at several shallow and deep sites within the study area.
7. Plants may be identified on site or they may be retained for later identity verification. Collected plants should be placed in sealable plastic bags, along with a label providing all

pertinent information. Sample collections should also be recorded in a notebook or on field sheets, along with a sample number, all pertinent site information and GPS location.

8. Samples to be archived or included in reference collections should be properly preserved in a plant press.

The survey data can be used to identify and describe plant communities or bed types. A study area reconnaissance in combination with a number of representative survey transects, facilitates a reasonable qualitative description and delineation of bed types and distribution. The end products for a qualitative Line Intercept survey include a map showing the distribution of plant bed types, and a list of species for each plant bed type and the water body or study area as a whole.

The preceding methodology describes a qualitative survey. Semi-quantitative results can be obtained through modification of the study design and the field sampling procedure as follows.

1. Select transect locations on a stratified-random basis, ensuring that all strata (i.e., bed types or geomorphically similar units) are equitably represented.
2. Conducting semi-quantitative sampling at 1 m intervals is likely to be prohibitively time consuming. Select larger intervals (e.g., 5 m or 10 m), or base sampling site selection on changes in bed type or a geomorphic characteristic(s) such as depth or substrate type.
3. Conduct sampling at each sampling site in a consistent manner and with a consistent level of effort. At each site, the same number of rake drags or rake tosses should be made and, to the extent possible, each drag or toss should sample an equivalent area.
4. At each sampling site, determine and record the relative robustness of plant growth. Example descriptors could include Dense, Moderate, Sparse and Trace.
5. For each rake sample, identify the species present and estimate the relative proportion of each species in the sample. Species that are present in very small amounts, perhaps only as fragments, can be recorded as 'present' or 'trace' only.

The methods described above are most effective where plants are readily visible from a boat, generally less than 1-2 m of depth. In cases where the littoral zone extends to greater depths Line Intercept surveys may be conducted using a snorkeler or divers, although such surveys are more difficult to conduct. If divers are used, they should be familiar with *in situ* identification of aquatic macrophytes, and must be certified (e.g., PADI, NAUI or equivalent) and familiar with accepted safe diving practices and department policy.

Transect with Quadrat

Determination of biomass is a quantitative assessment requiring considerably greater effort than qualitative assessment of aquatic macrophyte communities. Quantification of aquatic macrophytes is based on stratified-random sampling designs requiring at least a basic understanding of water body geomorphic conditions, and the nature and distribution of plant community types. Because of the effort involved, biomass studies are usually impractical in large waterbodies and are typically limited to smaller ponds, or discrete portions of lakes and rivers (e.g., bays or reaches).

1. Select transect locations on a stratified-random basis, ensuring that all strata (i.e., bed types or geomorphically similar units) are equitably represented.
2. Sampling sites may be pre-selected or field-selected, provided that they conform to study design protocols.
3. Conduct sampling at each sampling site in a consistent manner and with a consistent level of effort. The appropriate number of replicate samples collected at each site is determined in the study design phase.
4. At each sampling site, drop or throw the quadrat in a random manner and allow it to sink to the bottom.
5. A diver harvests all the plants rooted within the quadrat. Plants are cut at the water/substrate interface and the entire plants are placed in a mesh bag. Note: If study design requires that roots be collected as well, they should be dug out of the substrate and rinsed before placement in the bag.
6. Plants are brought to the surface and transferred to a plastic bag along with a label providing all pertinent information. Sample collections should also be recorded in a notebook or on field sheets, along with a sample number, all pertinent site information and GPS location.
7. Water depth should be recorded at each site. GPS waypoints, Secchi depth, and turbidity and bottom light level (if equipment is available) should also be recorded. Water temperature or temperature profiles should be recorded at several shallow and deep sites within the study area.
8. As soon as is convenient, plants from each sample are sorted by species. Whole plants and plant fragments are included, but senescent plants are excluded.
9. Each species is spun in a centrifuge dryer to remove all surface water. Plants are then weighed to determine the 'fresh' weight for each species within the sample. Total weights for each species are recorded and plants may then be discarded or re-bagged for further analysis.
10. Representative samples for each species should be retained for verification of species identification. Sample collections should be recorded in a notebook or on field sheets, along with a sample number, all pertinent site information and GPS location.
11. Samples to be archived or included in reference collections should be properly preserved in a plant press.

Biomass, or standing stock, is expressed as *fresh weight of each species* g/m^2, and *total fresh weight* g/m^2. Alternatively, biomass can also be expressed as *dry weight* g/m^2. Detailed procedures for determining fresh weight and dry weight are provided below.

Fresh Weight - Spinning Method

1. Place an individual sample into the centrifuge dryer (e.g., salad spinner) ensuring that any rocks, sticks and detritus are removed.

2. Centrifuge the sample for approximately one minute at a moderate speed (approximately 1 revolution per second). Appropriate spinning time will vary according to sample size and plant type, but samples should be spun until all the surface moisture has been removed.

3. Remove and weigh on an electronic balance. Alternatively, weigh with a spring scale in a plastic bag, subtracting the bag weight.

4. Record the weight.

Dry Weight - Oven Drying Method

1. Wash the sample using a sieve tray to remove rocks, debris and invertebrates.

2. Place each sample on a pre-weighed oven pan.

3. Label it and place it in an oven at 105 °C for 24 hours.

4. After 24 hours, remove and weigh the sample on a balance (remember to subtract the pan weight).

5. Record the weight.

Fresh Weight - Dry Weight Conversion

Specific study designs may specify use of fresh weight or dry weight, or both. In studies where sample sizes are large (i.e., >200 g) it is often impractical to process and dry entire samples, and fresh weights are used instead. Conversely, dry weights are more consistent and accurate, and may be more appropriate for studies with small sample weights (i.e., <200 g).

Fresh weights can be converted to dry weights by developing a conversion factor. To determine a conversion factor, sub-samples are first fresh-weighed and then dry-weighed. It is important to note, however, that fresh weight to dry weight correlation varies according to plant species and water body. Therefore, location-specific and species-specific conversion factors must be calculated for each survey and periodically verified.

Reference

Survey Methods

Survey methods were developed from information obtained from: Indiana Department of Natural Resources; Madsen (1999); Madsen and Bloomfield; and Parsons (2001)

Aquatic Macrophyte Identification

Useful aquatic macrophyte identification keys are provided by: Brayshaw (1989); Burland (1989); Brayshaw (2000); Crow and Hellquist (2000a); Crow and Hellquist (2000b); Kershaw et al. (2001); and Lahring (2003).

Aquatic Macrophyte Preservation

Methodologies for the preparation and preservation of aquatic macrophytes are provided by: Haynes (1984); Warrington (1994); and Parsons (2001).

2.14 Benthic Invertebrate Sampling Methods

Purpose

Open water benthic invertebrate sampling involves the collection of invertebrates that inhabit the upper sediment layers and the sediment surface. Typically macroinvertebrates are sampled with some meiofauna including early insect instars. Meiofauna is defined as microscopic animals that pass through 500 µm screen but are retained by a 64 µm screen. The mesh size used to collect/process the invertebrate sample determines the composition of the benthic invertebrate sample collected. Lakes and reservoirs are predominantly depositional environments and so Ponar or Ekman grab samplers are the most appropriate. Ponar grab samplers are most efficient at sampling harder sediments while Ekman grab samplers are most efficient in softer sediments. Open water benthic invertebrate sampling programs are usually conducted in early spring or late fall, when benthic communities tend to be the most stable. It is also important to maintain consistency of time of sample collection within and between years.

General

- Choose specific sampling locations at each site so that similarities in substrate type and sampling depth among sites are maximized.
- At any particular site, water sampling should be conducted prior to benthic invertebrate sampling to avoid disturbance of overlying waters by the sediment sampling technique.
- If the jaws are not closed properly when the dredge is retrieved, discard the sample and re-sample.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Ponar or Ekman grab sampler, rope (non-twisting nylon).
- 250 or 210 µm mesh sieve box.
- Labelled 1-L Nalgene® bottles
- Shoulder length rubber gloves
- Buffered Formalin (see Appendix C)
- Labelling tape, waterproof pen and note pad.
- Hydrographic map showing sample sites.
- GPS unit, depth sounder, tape measure, camera.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals
- Plastic pail, spoon and ladle.

Procedure

The procedure for sampling benthic invertebrate communities in lakes is similar to that described for sediment sampling in lakes. The type of grab sampler used will depend on sediment conditions and study objectives.

1. Label the sample containers with site identification, sample type, sampling method, sampler ID and the date of collection. Take a photograph of the site.
2. Note the following site information in the field sheet/book: target and actual sampling location (GPS); date and time of sample collection; overlying water depth (m); ambient weather conditions; grab penetration depth, depth sub-sampled, sampling personnel; any deviations from the field sampling procedure (FSP), macrophyte growth.
3. Ensure that the dredge jaws open and close properly.
4. Lock the dredge jaws in the open position and lower in a controlled fashion to the lake bottom. Do not allow the sampler to "free fall" The sampler should be in contact with the substrate or positioned just above it
5. Drop the messenger (if applicable) and slowly then quickly raise the dredge to the surface.
6. The sample is deemed acceptable if the desired depth of penetration has been achieved; and the sampler has completely closed and was not inserted on an angle or tilted upon retrieval. If the sample does not meet these criteria the sample should be retaken close to the original sampling location. The rejected sample should be discarded in such a way that it will not affect subsequent sampling efforts.
7. Place a container/bucket beneath the sampler just as it breaks the water surface.
8. Open the grab sampler over a 250 or 210 µm mesh sieve box. If substrate materials are predominately fines, gently wash the sample using the sieve box to release the fine sediment and transfer the contents retained on the sieve to pre-labelled 1 L plastic jar(s). Use more than one jar if the sample is large.
9. If substrates include significant amounts of coarse material or organic debris, on-site sieving may be impractical. In this case, samples may be double-bagged, labelled, kept cool and transported to a lab for sieving (i.e., with the aid of pressurized water). If samples can be kept cool and processed in a lab within a few days they may be preserved after sieving, otherwise the samples should be preserved at the time of collection.
10. Add buffered Formalin to the sample(s) to achieve a final concentration of 10%. If the sample contains a large amount of organic matter, algae and invertebrates, add approximately 1/5 the sample volume of buffered Formalin.
11. Add a waterproof label with the sample ID to each sample jar (in addition to an external label) and securely cap the jar(s). Agitate the jar(s) to ensure the Formalin is evenly distributed throughout the sample(s).
12. Rinse the grab sampler and the sieve in lake water to thoroughly remove residual sediment, invertebrates or plant material.
13. Site photographs may be taken if they aid in site characterization (e.g., nearshore sites or sites with aquatic plant growth). In addition, supporting data must also be collected to characterise the benthic habitat at that site.
    a) Water Depth: use a depth sounder to measure the water depth at the approximate location that the benthic invertebrate sample was collected.
    b) *In situ* Water Quality: Measure pH, DO, temperature and conductivity at the approximate location that the benthic invertebrate sample was collected, according to the multiprobe methods described in Section 2.1.
    c) Substrate Characterization: Collect an additional grab sample at each benthic invertebrate sampling location within the lake. If the sediment sample contains a lot of excess water, place the sample in a pail and carefully decant off as much water as possible without compromising the silt and sediment content. Stir (homogenize) the

sediment for 30 sec, then transfer into the appropriate pre-labelled containers for subsequent particle size analysis (grain size) and total organic carbon content. Store and transport samples in a closed cooler samples at 4 °C and do not allow to freeze.

d) Macrophyte cover: Where applicable, estimate % macrophyte cover at the sampling location and if possible list dominant species.

Reference

This protocol was derived from Alberta Environment (2002), Rosenberg et al. (2003), Environment Canada (2004) and RAMP (2005).

3.0 RIVER AND STREAM PROTOCOLS

3.1 Multiprobe Measurements, Long-Term Deployment of Dataloggers and Automated Sampling Equipment

Purpose

In situ measurements of parameters such as pH, DO, temperature, conductivity, turbidity and redox potential (when required) are routinely taken manually at the time of sampling in rivers and streams and also automatically over time (long-term deployment). Typically measurements using electronic single or multiprobe meters are taken at mid-depth in rivers and streams unless the river is particularly deep, then depth profiles may be taken. This sub-set of water quality parameters are best measured *in situ* because some parameters can only be measured *in situ* (i.e., temperature) or they have the potential to change during shipping to the laboratory (especially if the shipping time exceeds 24 hours; i.e., pH, turbidity). Long-term deployment of a Hydrolab® multiprobe allows measurements to be taken as a time series. This is useful for this sub-set of water quality parameters because several of these parameters vary over diurnal or daily time scales (e.g., pH, temperature, DO).

This section does not represent a comprehensive guide to the use of automated water quality monitoring equipment, for further information please refer to BC-MWLAP (1999) and Appendix D.

General

Taking accurate *in situ* measurements depends on strict adherence to calibration, maintenance and QA/QC procedures for the electronic water quality meters used. Maintenance and calibration should be carried out according to the manufacturer instructions and additional technical assistance can be found on the internet and through consultation with technical representatives. The manufacturer manuals are not necessarily comprehensive, so an additional information compilation exercise through these sources is often worthwhile. Maintenance and calibration log-books should be kept up to date to track the performance of the meter. The meter probes should be calibrated daily under field conditions and temperatures, and periodically throughout the day if required due to intense use under extreme conditions (e.g., DO at sites of different altitudes or every five samples if water quality changes dramatically from site to site). The exceptions are: temperature (check in lab monthly with a certified mercury thermometer), conductivity and turbidity (calibrate at the beginning of the sampling trip), and redox potential (calibrate once every six months). Always calibrate the pH meter with at least two buffer solutions (either a combination of: pH 7 and pH 4 (sample pH <7) or pH 10 and pH 7 (sample pH >7)). Consult the manufacturer manuals and available information for the meter for specific calibration information.

Review the water quality data on-site during sample collection to prevent the measurement and/or recording of false measurements. Measure again and double check any dubious readings before leaving the site. It may also be worthwhile doing verifications at the end of the day for some key parameters (not re-calibrations) to check if the meter has drifted or is malfunctioning. Meter readings should be checked in standard solutions and recorded in the log/field book to ensure that the meter has been working properly throughout the day.

Service the probes on a regular basis (every two to three weeks) and keep records of calibration and maintenance.

Equipment

- Single or multiprobe electronic water quality meters from reputable brands (e.g. YSI®, Horiba® or Hydrolab®, etc). The recommended minimum accuracy for the field measurements is: DO (± 0.2 mg/L); temperature (± 0.4 °C); conductivity (± 0.05 µS/cm); pH (± 0.02 units); turbidity (± 0.01 NTU); and salinity (± 0.1 ppt) (RAMP 2005).
- Standard calibration solutions (within the expiry date).
- Field log book and calibration log sheet/book (waterproof paper) (an example of a river field data sheet is in Appendix F).
- Lint free wipes (e.g., Kimwipes®), tap water, distilled water.
- Barometer and thermometer.
- Winkler equipment (see Section 3.2).
- Pelican® case to store the meter when not in use.
- The meter manual and any items required to change membranes or perform minor multiprobe repairs.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals (e.g., calibration solutions).

Procedure

In situ Field Measurements

1. Assemble the meter according to the instruction manual.
2. If sampling:
   a) by wading - place the connected sonde, complete with circulation, in the main flow with the probes oriented upstream to dislodge air bubbles.
   b) low bridge/slow flow - lower the sonde carefully into the main flow.
   c) high bridge/fast flow - collect a full stainless bucket of water from the main flow - raise carefully to bridge deck and place sonde in the pail, ensuring unit is circulating. If you are using a Hydrolab® and the circulator will not fit, constantly swirl the sonde in a figure eight pattern until readings stabilize and are recorded. Take readings as quickly as possible to avoid temperature and oxygen changes.
3. Hold the sonde unit below the surface until the DO reading stabilizes. Ensure unit is circulating.

4. Record temperature, pH, conductivity, DO and % saturation on the field sheets under the appropriate "bank" column (left, center, right). Left and right bank is determined with the sampler facing downstream.
5. Calibrate the multiprobe/meter prior to daily sampling for pH, DO, conductivity and turbidity. Redox potential should be calibrated every six months. Temperature should be checked every month with a certified mercury thermometer.
6. *In situ* measurements should be taken just below the surface of the water (0.1 m depth). In deep rivers they should also be taken at 1 m intervals down to 1 m above the bottom. At depths of ≤2m one set of measurements at mid-depth may be considered to be appropriate. At depths between 2 and 4 m measurements can be taken 0.25 m above the river bottom. In the case of deep rivers, the probe may require additional weight to ensure a vertical profile. Important: Additional weight should never be attached directly to the probe or cable. A weighted line may be tied or taped to the probe cable, and the strain of the assembly weight should be borne by this line at all times to avoid damage to the probe cable.
7. Let the instrument stabilize at each depth (may require 1-2 min) and record the readings on a field sheet/book. Also, if possible store readings at each depth in a datalogger. On deep profiles, it may be acceptable to proceed at 5 m intervals when there is little change in readings at 1 m intervals if agreed to by the project manager. When change is detected (thermocline, chemocline etc.), then define the area of change at 1 m intervals.
8. Bring the probe/sonde back up to 1 m, allow it to stabilize and record the readings. (Note: redox will probably not stabilize quickly at the surface.) This acts as a field check on the instrument and verifies the accuracy of the first reading.
9. One water sample taken at one profile depth per water body is subjected to a Winkler analysis as a further check of the accuracy of the meter measurement, preferably at a depth where oxygen appears stable. Meter DO measurements within ±0.5 mg/L of the Winkler DO measurements are considered acceptable.

Set Up and Long-Term Deployment of Hydrolab®Multiprobe Dataloggers and the ISCO® Automated Sampler.

Protocols for the set up and long-term deployment of some Multiprobe Dataloggers and the Automated Sampler are given in Appendix D. There are other multiprobe dataloggers commercially available but Hydrolab® dataloggers have been shown to be reliable and are among the most commonly used. Alberta Environment currently uses Hydrolab® and YSI® multiprobe dataloggers and the ISCO® Automated Samplers in their automated water quality monitoring.

Reference

This protocol was derived from Environment Canada (1983), Alberta Environment (2002), Environment Canada (2004) RAMP (2005), USGS (2005), EMAN-North (2005).

Further information regarding *in situ* field water quality measurements can be found in USGS (2005). For further reading on the use of automated water quality monitoring equipment please refer to BC-MWLAP (1999).

3.2 Winkler Dissolved Oxygen - Azide Modification

Purpose

The iodometric (Winkler) titration procedure is not routinely used to determine the DO status of a water body, rather the Winkler procedure is used to confirm electronic meter readings and calibrations. The Azide Modification of the Winkler method described is the standard analytical test for DO. This method is not generally recommended for *in situ* routine DO measurements because it is time consuming, the potential interference from nitrite and iron in the water may decrease the accuracy of the technique, and preventing exposure of the water sample to atmospheric oxygen in the field can be difficult.

General

- Replace the rubber tubing in buckets yearly.
- Test the DO buckets for leaks by timing the fill time of each bucket. Realign the lids or replace gaskets if necessary.
- During cold weather, keep the tubing and holes free of ice crystals. Transfer full bottles to a warm cooler quickly to avoid freezing and breakage. Never set full Winkler bottles on the ice as the bottoms of the bottles may break due to the unequal rate of expansion/contraction of water and glass.
- Do not acidify until 5 min before titration, especially on river samples containing pulp mill effluent.
- Final values may be averaged or entered as replicates depending on the project requirements.
- See Appendix C for Reagent Preparation.
- Caution is required when titrating stained water, samples very low in oxygen or turbid samples. The endpoint can be difficult to detect and easily exceeded.
- HACH powder pillows may be used, but the replication and accuracy of these samples may be unreliable.

Equipment

- BOD bottles (300 ml) complete with stoppers
- DO bucket and rope
- Manganous sulphate and alkaline iodide azide solutions (made at McIntyre Lab)
- Concentrated H_2SO_4, starch solution,
- 250 ml Erlenmeyer flasks, burette
- 0.025 N sodium thiosulfate (standardized weekly)
- BOD bottle containing preserved sample
- Stir plate and stir bars
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals.

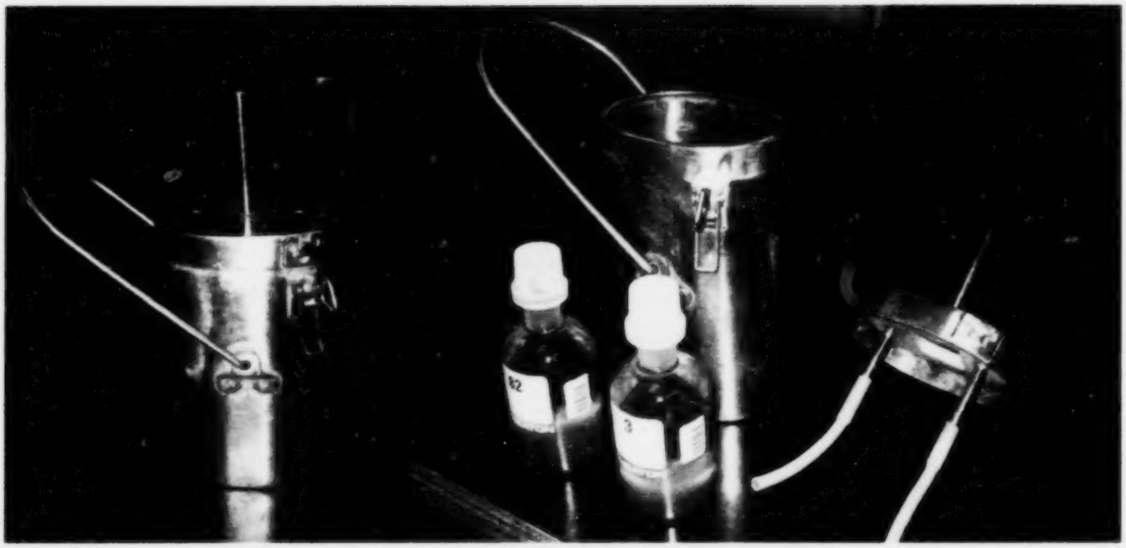

Figure 3.2-1 **Winkler bucket assembly**

Procedure

Sampling

1. Record the bottle numbers on field sheet.
2. Place two empty BOD bottles in the DO bucket and replace the lid so the EXTENDED tubing is positioned inside each bottle. Make sure the lid has a good tight seal and no air escapes from around the rim when lowering bucket into water.
3. Lower the DO bucket directly into the flow.
4. Hold the bucket underwater until air no longer escapes from the metal tube (if sampling under ice, ensure that the bucket is below the ice level).
5. Remove the bucket from the water and stopper bottles immediately.
6. Remove bottles from bucket. Add 2 ml of manganese sulphate, then 2 ml of alkaline iodide azide in quick succession then stopper it. Invert rapidly several times. Immediately repeat treatment to second bottle.
7. Invert both bottles fifteen times then place protective plastic cap over each stopper.
8. Resample if large bubbles are present in the bottles.
9. Keep the samples cool and in the dark. Titrate within 24 hours. Normally, the water quality meter DO value should be within ±0.5 mg/L of the Winkler DO value.

Reference

This protocol was derived from Alberta Environment (2002) and USGS (2005).

3.3 Grab and Integrated Water Sampling

Purpose

There are two ways to take a representative water quality sample from a stream or river, by taking a grab sample (discrete or composite), or by taking a depth integrated sample. A grab sample is a sample either taken by hand, or using sampling equipment (e.g., Kemmerer grab sampler, pumping system), from a discrete depth in the water column often 30 cm below the water surface. A discrete grab sample is taken from one location in a river or stream, whereas a composite grab sample is the combination of a number of discrete grab samples taken from several locations in the stream or river. Often discrete grab samples are taken from streams and smaller rivers, whereas composite of grab samples are taken from larger rivers, at several locations on a transect crossing the width of the river at a particular site. Discrete grab samples may be collected across the channel of a stream where the spatial variability is of interest (e.g., in an effluent plume). Conversely, grab samples may be collected across the channel and composited if the spatial variability is not of primary interest but the overall conditions are (i.e., the 'average' condition across the channel), and it isn't practical to sample at the fully mixed location.

Similarly, depth-integrated samples are taken in rivers where water quality may vary with depth. The size of the river or stream, the study objectives and the sampling effort allocated to the study will determine the type of samples taken. Samples should be taken from sites progressing in order from the least contaminated site first to the most contaminated site last unless a time of travel study is undertaken. The degree of contamination can be estimated from historical data, site conditions, land use, professional knowledge etc.

General

- Only use sample bottles provided by the analytical laboratory specific to each analysis. Reject any uncapped bottles (especially those for analysis of trace metals and other contaminants). Ensure there is always at least one extra set of bottles on hand.
- Ensure bottles remain capped until sample collection and are stored under clean conditions (e.g., in cooler, plastic bag etc). Vehicles should also be kept reasonably clean to limit potential contaminant sources.
- Only leave the sample bottle uncapped while filling the bottle and/or adding preservatives. Do not touch the cap liner or the inside of the sampling bottles (even while wearing gloves). Only the water sample and the preservative should touch the inside of the sampling bottle or the cap.
- Review of documentation accompanying the sample bottles referring to sample collection, storage and transport, and consult with laboratory personnel regarding these requirements. If samples are to be submitted close to or on the weekend, make arrangements with the laboratory to ensure sample holding times are still enforced.
- Ensure all preservatives are sealed and within the marked expiry date. Add preservatives to samples in an area away from potential sources of contamination (e.g., roads and car

parks [dust and hydrocarbons]). Read the MSDS sheets for all preservative chemicals and wear safety glasses and gloves while preserving the samples.

- Samples should always be collected at the bow of the boat because the bow could potentially be pointed up stream if it is anchored at the bow, reducing the potential for contamination from the boat or motor. Ensure sample is collected in good flow, not in an eddy or backwater. When grab sampling, face upstream to avoid any stirred-up sediments and sample 30 cm below the water surface.
- Colour code sample bottles to ease collection and prevent mix ups.
- Keep all sample collection equipment in a sealed clean plastic bags or in a clean cooler when not in use to prevent contamination.
- Sampling personnel should wear unpowdered latex or polyethylene disposable gloves while collecting water samples and refrain from smoking or eating. Do not use insect repellent if sampling by hand or be very careful.
- Take a photograph and GPS coordinates at each site and follow procedures outlined Section 2.1 for taking *in situ* water quality measurements. Record observations of the sampling site.
- Sample tributaries well upstream of the main river to avoid mixing zones.
- River size determines whether grab, vertical integrated, composite or transect samples are collected. Review with project manager.
- See Section 4.2 for specific guidelines related to trace organic sampling in rivers.
- See Section 4.3 for sampling equipment cleaning and decontamination procedures.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Sampling container - 10-20 L capacity, such as a large carboy complete with lid (material of container will depend on variables of interest). If sampling for chlorophyll, a light proof container is required (e.g., use a black garbage bag to cover more transparent containers).
- 1L sample bottle, foam ring, bungee cord
- Trace organic sampling equipment: Stainless 4 L bottle holder with insert for holding trace organic bottles (with rope) (Figure 3.3-1), stainless steel 20 L bucket (long rope for bridge sampling)
- Winkler sampling equipment: Winkler bottles, chemicals, DO bucket and rope
- Sample bottles and preservatives in coolers plus an extra set of sample bottles and preservatives. Ice packs and hot water bottles, depending on season
- 10 L carboy of deionised water. Use fresh deionised water (do not store for extended periods of time; do not use after 6 months)
- Lab analysis request sheets and/or Chain of Custody forms (COCs)
- Ziploc® bags, labelling tape, waterproof field sheets and markers/pens, garbage bags.
- GPS unit, depth sounder, camera, current or flow meter, waders.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Equipment for field cleaning/decontamination procedures (see Section 4.3)
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals

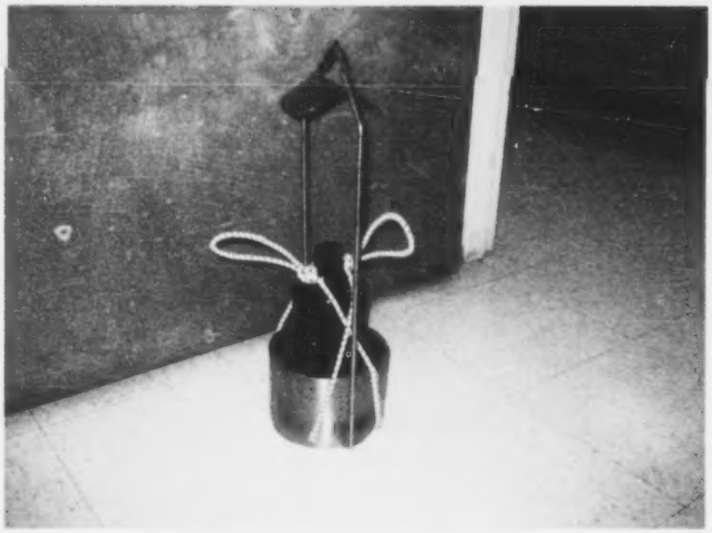

Figure 3.3-1 **Stainless steel 4L bottle holder with insert and trace organic bottle**

Procedure

Discrete Grab Sampling

1. Sample containers should be rinsed three times with ambient water prior to final sample collection. **Note: Do not rinse all sample bottles**. Rinse all sample bottles and caps with water from the appropriate depth three times before filling them, except for those destined for:
   a) bacteriological analysis, synthetic organic compounds, pesticides;
   b) volatile hydrocarbons (e.g. including total volatile hydrocarbons [TVH], total extractable hydrocarbons [TEH], benzene, toluene, ethylbenzene, and xylene [BTEX]); and
   c) total petroleum hydrocarbons (e.g., TVH, TEH, and BTEX), oil and grease. It is recommended that sample bottles for these parameters should not be rinsed because some parameters can accumulate on the container during rinsing or bottles contain preservative. Also, in the case of bacteriological analysis, rinsing was not recommended in the interest of maintaining sterile conditions.
   d) suspended solids, parameters associated with suspended solids, and
   e) trace metals (although some laboratories may still request that trace metal bottles be rinsed.

   Check with the project manager and the analytical laboratory as to whether sample bottles subject to the analyses described in 3d and 3e should be rinsed. The reasons given for not rinsing sample bottles for the analyses listed in 3d and 3e are: some parameters can accumulate on the container during rinsing and sample containers have already been certified to be free of contaminants by the analytical laboratory. However, it is particularly important to make sure these sample bottles are received capped and remain capped until sampling.

2. Submerge bottle to a depth of approximately 30 cm, uncap and fill bottle, and recap at depth (to avoid contamination). At the surface, remove the lid and pour the water out away from the sampling area. Repeat this procedure twice more before collecting the sample.
3. Direct fill sample bottles approximately 30 cm below the surface. While sampling avoid submerged vegetation and ensure sample is free of obvious foreign material not representative of the water column at time of sampling (e.g. algae, sediment, organic matter etc.). Cap tightly and place into cooler for transport.
4. If the ultra-clean techniques (e.g., "clean hands/dirty hands") are required to collect samples for some ultra-low level analyses then sampling/handling instructions provided by the laboratory should be followed (e.g., mercury).
5. Immediately after collection store the water samples at 4 °C in a closed cooler. Do not allow to freeze.
6. Filter and/or add preservatives to appropriate bottles, immediately, or as soon as possible after sample collection. If the samples are to be laboratory-filtered, ship them as soon as possible and ensure they arrive in the laboratory well within the specified hold time for un-filtered and un-preserved samples. Unfiltered chlorophyll-a samples should be stored in a dark bottle to minimize light exposure.
7. All sample bottles should be clearly labelled with date, location, site, depth, analytical parameter group and sampler ID. Store and transport all samples at 4 °C in a closed cooler. Do not allow to freeze.

Composite Grab Sampling

The Regional Aquatics Monitoring Program (RAMP) collects grab water samples from a river to form a composite sample, using the following wetted width designations:

- Wetted width >50 m: Three grabs at each of five equally spaced sample locations along river cross-section.
- Wetted width 20-50 m: Four grabs at each of three equally spaced sample locations along river cross-section.
- Wetted width <20 m: Ten grabs from a single centre-channel position.

The number of grab samples to be taken at a particular river site should ultimately be determined by the project manager. In deeper rivers, discrete depth samples may be taken using a Van Dorn/Kemmerer sampler or a GeoPump® sampler, instead of surface grab samples for preparation of a site composite sample.

Where the spatial composite sample is comprised of grab samples taken 30 cm below the water surface, the following procedure should be adopted for grab sampling at each location.

- An intermediate sample bottle should be rinsed three times with ambient water prior to final sample collection. Submerge to a depth of 30 cm, uncap and fill bottle, and recap at depth (to avoid contamination).
- At the surface, remove the lid and dispense away from the sampling site. Repeat this procedure twice more before filling the container at 30 cm below the surface and recapping at depth. While sampling avoid submerged vegetation and ensure sample is

free of obvious foreign material not representative of the water column at time of sampling (e.g. algae, sediment, organic matter etc.).

- Pour the water sample into the composite sample bucket and continue to sample until sufficient composite volume has been collected (do not rinse again). Keep the bucket covered during sampling.

Sub Sampling Procedures

1. Cap and shake the sample container well (approx. 30 sec.) before pouring each sample.
2. Minimize dust contamination by pouring samples in a calm area and by capping sample bottles immediately before and after filling. Do not touch the inside of the sample bucket, bottle lids or mouths with your hands. Chlorophyll-a samples should be poured neither first nor last.
3. Note: Do not rinse all sample bottles. See rinsing instructions given in Step 1 in the Discrete Grab Sampling protocol.
4. Hold the sample bottle caps lid-down while filling the sample bottle. If the ultra-clean techniques (e.g., "clean hands/dirty hands") are required to collect samples for some ultra-low level analyses then sampling/handling instructions provided by the laboratory should be followed (e.g., mercury).
5. Immediately after collection store the water samples at 4 °C in a closed cooler. Do not allow to freeze.
6. Filter and/or add preservatives to appropriate bottles immediately or as soon as possible after sample collection. If the samples are to be laboratory-filtered ship them as soon as possible and ensure they arrive in the laboratory well within the specified hold time for un-filtered and un-preserved samples. Unfiltered chlorophyll-a samples should be stored in a dark bottle to minimize light exposure.
7. All sample bottles should be clearly labelled with date, location, site, depth, analytical parameter group and sampler ID. Store and transport all samples at 4 °C in a closed cooler. Do not allow to freeze.

Depth Integrated Sampling

The following procedure describes the use of a depth sampler constructed by Alberta Environment to collect a vertical integrated sample from rivers. It is also possible to use Van Dorn/ Kemmerer depth samplers or Peristaltic pump tubing, utilising the sampling procedures outlined in Section 2.4.

1. Place 1 L sample bottle into foam ring or 4 L jug into stainless steel holder (Figure 3.3-2) and secure using bungee cord.
2. Lower the bottle, in the stainless steel holder, off the bridge or under the ice.
3. Rinse the sample container three times with sample water.
4. To obtain water sample, slowly raise and lower the bottle through the water column until full.
5. Do not contact the river bottom.
6. Cap and shake the bottle vigorously before each sample is poured off.
7. Rinse and fill sample bottles as described for the composite sub-sampling technique in streams and rivers (previous section).

8. Immediately after collection store the water samples at 4 °C in a closed cooler. Do not allow to freeze.
9. Filter and/or add preservatives to appropriate bottles immediately or as soon as possible after sample collection. If the samples are to be laboratory-filtered ship them as soon as possible and ensure they arrive in the laboratory well within the specified hold time for un-filtered and un-preserved samples. Unfiltered chlorophyll-a samples should be stored in a dark bottle to minimize light exposure.

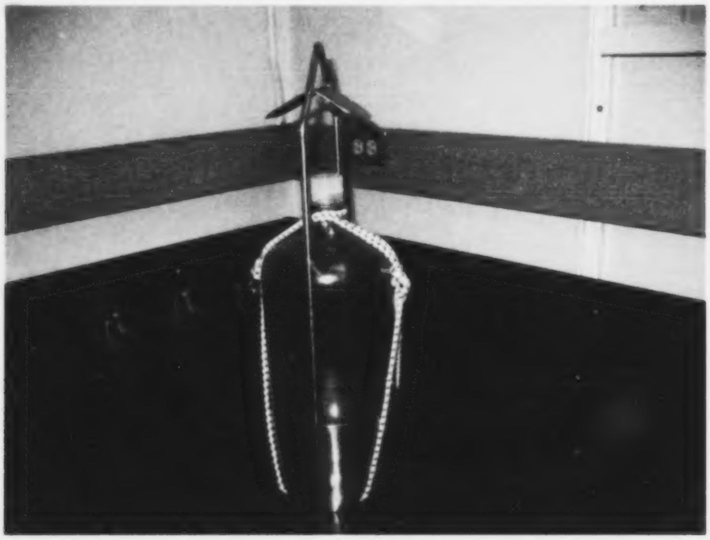

Figure 3.3-2 **Stainless steel 4L bottle holder with 4L jug**

Reference

This protocol was derived from Environment Canada (1983), BC-RISC (1997a), Alberta Environment (2002), RAMP (2005); and EMAN-North (2005).

3.4 Under-Ice Water Sampling

General Considerations

- Sampling locations to be sampled in the winter should be located as close as possible to the open-water stations
- Clear loose ice and snow; drill through the ice with a hand or motorized auger. Keep the area around the hole clear of potential contamination (e.g., dirt, fuel, oil, etc.). Avoid gas, oil and exhaust contamination of the sampling equipment.
- Remove all ice chips and slush from the hole using a plastic sieve.
- Samples should be collected approximately 0.2 m below the bottom of the stream ice using a depth sampler (e.g., Van Dorn sampler) or a GeoPump® sampler to minimize the possibility of contaminant introduction associated with augering if a motorized auger is used. Don't let the sampler come into contact or stir up sediment from the stream bottom as this will contaminate the water sample.
- In extreme cold temperatures well below freezing, do not rinse the sample bottles, the rinse water will freeze to the surface of the bottle – consult project manager for alternate protocol.
- Otherwise follow the sampling procedures outlined for open water sampling using the GeoPump® and depth samplers.
- Record ice depth and total depth

All sample bottles should be clearly labelled with date, location, site, depth, analytical parameter group and sampler ID. Store and transport all samples at 4 °C in a closed cooler. **Do not allow the samples to freeze.**

Reference

This protocol was derived from Environment Canada (1983), BC-RISC (1997a), RAMP (2005) and EMAN-North (2005).

3.5 Bacteriological Sampling

Purpose

Bacteriological samples are taken to assess the microbiological and sanitary quality of the water and to assess the potential health risk from waterborne diseases. Samples are typically analysed for a combination of the following bacterial parameters: total and faecal coliforms, *Escherichia coli (E. coli)*, fecal streptococci and enterococci. Due to the high risk of potential contamination of the sample during collection, care must be taken when collecting bacteriological samples. Hence, additional procedures are followed to try and maintain sterile conditions.

General

- Keep the sample at 4 °C and deliver it to the Provincial Laboratory of Public Health within 24 hours. Do not freeze.
- Affix the request sheet number to the bottle upon return to the office.
- Complete the Request for Bacteriological Examination of Water form ensuring that the form and bottle number are identical. Ensure that the sample number, site name, date, and time are clearly marked on the sheet. Time is recorded in MST.
- Samples are not accepted by the lab after noon on Friday, or weekends, unless special arrangements are made as media must be prepared (especially for E. coli).
- When collecting more than a few samples, contact the Provincial Lab prior to the sampling trip and let them know the number and type of samples to be collected.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Sterile bacteriological bottles supplied by the analytical laboratory.
- Sample request sheets and Chain of Custody forms (COCs)
- Depth sampler (e.g., Van Dorn, Kemmerer) - for deep water sites
- Labelling tape and waterproof markers and field sheet/book
- GPS unit, depth sounder, camera, tape measure.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).

Procedure

Grab Bacteriological Samples

1. Always hold bottle upright and by the base, as the preservative is already in bottle.
2. With the bottle mouth directed toward the current, fill the sample bottle to the 200 ml line, with the bottle opening 30 cm below the surface. Immediately cap the bottle securely. Place the bottle immediately in a closed cooler with ice packs or hot water bottles, depending on the season.
3. If necessary, the sample bottle can be filled from a sterile intermediate container.

Offshore Sampling at Depth

1. Collect a sample of water at the desired depth with a depth sampler according to Section 3.3.
2. Do not rinse the bottle or touch the inside of the bottle or cap, and always hold bottle upright and by the base, as preservative is already in bottle. Keep sample bottle closed until needed.
3. Fill the sample bottle to the 200 ml line and immediately cap the bottle securely. Place the bottle in a closed cooler immediately with ice packs or hot water bottles, depending on the season.

Reference

This protocol was derived from BC-RISC (1997b), Alberta Environment (2002), USGS (2005), Calgary Health Region (2005) and EMAN-North (2005)

3.6 Protozoan Sampling

Purpose

Similar to bacteriological sampling, protozoan samples are taken to assess the microbiological and sanitary quality of the water, and to assess the potential health risk from waterborne diseases. The predominant protozoan pathogens in aquatic systems are *Cryptosporidium* and *Giardia*, and the presence of these can only be verified by the identification of *Cryptosporidium* oocysts and *Giardia* cysts. Due to the high risk of potential contamination of the sample during collection, care must be taken when collecting protozoan samples. Thus, additional procedures are followed to try and maintain sterile conditions. The sampling protocol described in the following sections relates to sampling raw and finished water from surface water sources. For influent and effluent sampling protocols refer to Alberta Environment (2002).

General

- When sampling additional sites on the same day, approximately 25 ml of Neutrad® should be added to the intake line at the strainer when the flushing phase started at the next site.
- In addition, the meters occasionally plug up or stop working during high turbidity conditions. When this occurs and it is not possible to clear the meter or replace it, a 10 L plastic carboy can be used to collect the water from the effluent line in order to get a total volume of water pumped.
- When sampling raw water at the water treatment plants, the above procedure is followed but the Pony® pump is not used. Instead, the intake line on the sampling apparatus is connected directly to the raw water sampling tap at the plant.
- When sampling finished drinking water at the water treatment plants, the above procedure is followed with these exceptions:
- A separate filter apparatus with a venturi regulator valve is dedicated to this collection and not used elsewhere to avoid any contamination.
- A sample volume of 1000 L is passed through the filter.
- The intake line on the sampling apparatus is connected directly to the finished water sampling tap at the plant, thus eliminating the Pony® pump.
- To dechlorinate the drinking water, sodium thiosulfate is added to the sample via venturi, at a rate of 10 ml/min. The flow rate is adjusted using the calibration screw on the pressure regulator.
- The flow rates of the water, as well as the sodium thiosulfate are checked immediately and again periodically during pumping. The psi reading is also monitored to ensure pressure doesn't exceed 30 psi.

Equipment

- 12 volt battery
- 12 volt Pony® pump
- Sampling apparatus consisting of and assembled in the following order: strainer, intake hose, pump, flow regulator valve, pressure gauge, filter holder with inlet and outlet connector hoses, water meter, effluent hose (Figure 3.6-1)

- Cartridge filters, 10 in., 1 micron yarn-wound polypropylene
- Bucket, Ziploc® bags, Neutrad® soap, stopwatch
- 50 or 100 ml bottles for turbidity sample
- Cooler and ice packs or hot water bottles, depending on the season.
- Ice pick or suitable anchoring rod
- 1 L Nalgene® bottle, 10 L Nalgene® carboy
- Labelling tape, waterproof markers and field sheet/book
- Sterile bacteriological bottles supplied by the analytical laboratory
- Sample request sheets and Chain of Custody forms (COCs)
- Labelling tape and waterproof markers and field sheet/book
- GPS unit, depth sounder, camera, tape measure.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).

Additional materials for Finished Water sampling

- 2.5 litres 2% sodium thiosulfate solution
- 100 ml graduated cylinder
- Small screwdriver
- Thin flexible tubing
- Hollow ceramic weight

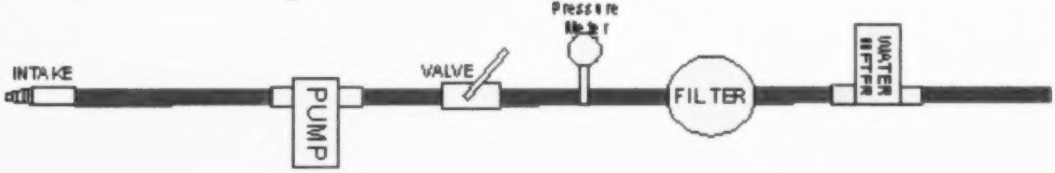

Figure 3.6-1 **Assembled protozoan sampling apparatus**

Procedure

Raw and Finished Water Method

Collection of raw water samples from surface water sources:

1. The sampling apparatus is set up on shore with the intake line placed in the water column midway between the surface and bottom and held in place using an anchoring rod. Care is used when choosing a site to find good flow and to avoid any backwater areas.
2. The pump is connected to the battery and the flow valve is opened for maximum flow. 100 L of source water are pumped to flush the system and check for leaks. The valve is shut off and the pump disconnected from the battery.
3. A cartridge filter is aseptically placed into the filter holder. The filter holder is tightened and placed in the stainless steel bucket to keep it upright.
4. A water meter reading and the time are recorded. The pump is started and the valve partially opened. A 1 litre Nalgene® bottle and stopwatch are used to adjust the valve to a flow rate of 4 L/min

5. At least 100 L of water are pumped through the filter if possible. In water that is very clear additional sample was pumped through (usually 150-200 L). In very turbid conditions pumping continues only until the filter has reached capacity and the water flow has ceased.
6. The pressure is monitored to ensure it does not exceed 30 psi. During pumping, the flow rate is adjusted when necessary to try to keep the rate constant.
7. When pumping is complete, the flow valve is closed to ensure no backflow of sample, and then the pump is disconnected from the battery. The end time and meter reading are recorded.
8. The filter is aseptically removed from the filter holder and put into a plastic Ziploc® bag. The water and any sediment from the filter holder are poured into the bag containing the filter cartridge, then the bag sealed and put into a second Ziploc® bag to ensure that any leakage will be captured.
9. Sampling information, including site name, date, start and end times, start and end meter readings, total volume pumped, flow rate, maximum pressure, sampler initials, and Envirodat sample number are recorded on the outside bag.
10. The sample is placed in a cooler containing ice packs or hot water bottles, depending on the season. If not transported to the laboratory the same day, keep refrigerated at 4°C at the office until they can be sent. A 50 - 100 ml water sample for turbidity is also collected whenever a filter sample is taken.

Cleaning of Sampling Apparatus

1. When sampling has been completed for the day, the sampling apparatus is attached to the Pony® pump and intake line, which in turn is attached to a water tap. A minimum of 100 L of warm water is flushed through the system. Neutrad® soap is added at the beginning of the cleaning. The filter holder is scrubbed with a stiff brush in instances where grit was present. Once washed, the equipment is left to air dry.
2. With the sewage sampling equipment, all gear is thoroughly cleaned with Neutrad® and hot tap water and left to air dry. Once dry, all equipment is stored in clean plastic bags.

Reference

This protocol was derived from Alberta Environment (2002).

More information on protozoan sampling procedure is available in USGS (2005).

3.7 Epilithic and Epipsammic Periphyton Sampling

In rivers and streams, benthic algal communities typically account for most primary productivity. Benthic algal communities living on substrate surfaces are collectively referred to as periphyton. Sampling protocols in this section are concerned with the quantitative assessment of two kinds of periphyton; epilithic periphyton are attached to the surfaces of rocks or other objects projecting above stream bottom, and epipsammic periphyton are associated with sand. There are two main components to this type of sampling:

- location of the sampling points along a transect in the stream or river; and
- collection of periphyton from the substrata

Epilithic periphyton communities can be sampled using a variety of methods but two recommended methods are described here: the template method commonly used by Alberta Environment, and the collar method. A method of sampling epipsammic periphyton communities is also described here.

Template Method for Sampling Epilithic Algae

Purpose

The template method is used to quantitatively sample epilithic algae for chlorophyll-a and Ash Free Dry Weight (AFDW) biomass determination and species identification. This method is used to sample the upper surfaces of stones (i.e., the area exposed to the direct stream flow). This helps alleviate the effects on sampling from spatial differences in water velocity, erosion of communities along the substrata edge, and grazing invertebrates that tend to inhabit the under surfaces or along the edges of stones. This method also remains effective in cases where there are large mats of long filamentous algae.

General

- Discuss the site selection and the collection and quality control procedures with the project manager before going into the field. Often shady areas of the river will be avoided.
- To further standardise sampling effort, sample a sub-set of pre-defined conditions (depths of 40 cm, velocities of 0.3-0.5 m/s, only cobble sized substrates, etc.). This may be particularly useful when control sites are quite different to impact sites. Discuss with the project manager.
- Sample in an area that will have flowing water covering the rocks throughout the open water season. Watch for changes of stage prior to sampling trip, as high stage might mean sampling recently exposed substrate.
- Ensure that the scalpel and template are cleaned between scrapes.
- Keep the algae out of direct sunlight.
- Record full description of the site - flow, % algae cover, description of algae, recent changes in water level, etc. Take photographs of each site from upstream and downstream directions.

- Keep all acids, including that from skin, away from rocks, filters, and any apparatus used as this can degrade chlorophyll. $MgCO_3$ can be added to neutralize any acids present in the sample but this optional.
- Invertebrates must be excluded (the large ones, to a reasonable extent) since they eat algae. Avoid leaving any small stones on the filter as rocks will damage the laboratory high speed homogenizer used to grind the samples, and could endanger the laboratory technician.
- Carefully lift off scrapes so algal cells are not destroyed.
- Where boulder/rocks are too big or are too embedded to be brought to the water surface, an underwater bedrock/boulder quantitative sampling technique should be considered (see Biggs and Kilroy [2000] for details).
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- 20-30 m tape measure.
- 2 pegs (>20 cm long) and mallet.
- Sample cooler and dry ice/ice.
- Scalpels (#3) & blades (#10).
- Site labels, waterproof pens, Ziploc® bags/Whirlpaks®.
- 4 cm^2 and 2 cm^2 flexible plastic templates, aluminium foil squares- 10x10 cm.
- $MgCO_3$ (powder), distilled water.
- Scintillation vials, Small plastic wide mouth jars, approx. 25 ml, Parafilm®.
- GF/C filters, forceps, turkey baster.
- Trays for rock collection (plastic ones from cooler or stainless steel, etc.).
- Clip and board to affix foil and filter to.
- Lugol's solution (see Appendix C).
- Labelling tape and waterproof markers and field sheet/book
- GPS unit, camera, tape measure.
- Disposable unpowdered latex, long rubber gloves.
- Safety equipment (see Section 4.4).

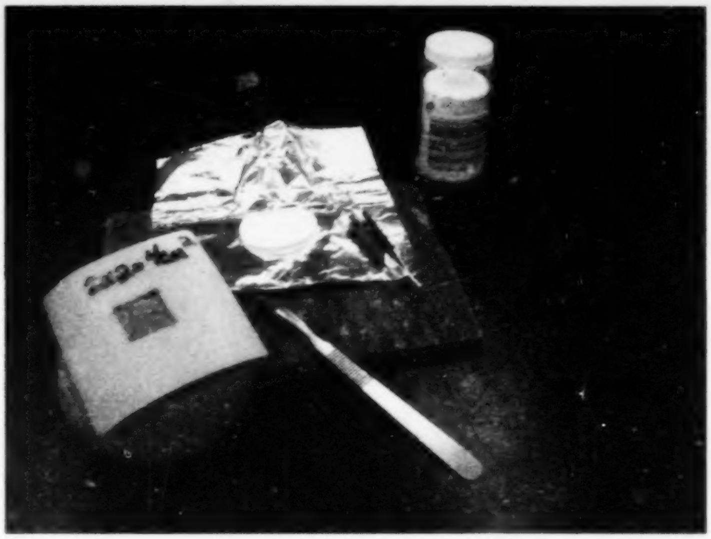

Figure 3.7-1 Example of a 4 cm^2 flexible plastic template

Procedure

Chlorophyll-a Sampling

1. Rocks to be sampled for periphyton should be sampled across a transect extending the width of the watercourse unless river is too deep. This can either be an imaginary transect or a defined transect. A transect can be defined in smaller watercourses by:
    - selecting a reference point in the middle of the site and driving a peg into the ground on one of the banks;
    - attaching a tape measure to the peg and laying it out taught across the watercourse. Anchor the far end with the second peg (other bank); and
    - divide the stream width into equally spaced intervals according to the number of rocks that are to be sampled (consult with the project manager).
2. It is important to be random in the rock selection. Wade along the imaginary transect out from shore or a rope can be stretched across river, taking 2 steps then select a rock (minimum size of 5 cm diameter) from approximately 40 cm depth. Long arm gloves can be worn for this. For the defined transect wade out to the first marked point and without looking pick up a stone. If the stone is <5 cm diameter or a sandy, silty areas between cobbles is touched, then take the nearest stone that is >5 cm diameter.
3. All the stones can be collected at once, or individually, before returning on the stream bank to sample. Place the stone(s) on a white tray with a small amount of stream water and return to the stream bank
4. If the river becomes too deep, head upstream repeating the above steps until all rocks are collected.
5. Orient each rock as it was in the river, and place the 4 cm^2 template over the area (chosen randomly) to be scraped. Only take a 2 cm^2 scrape per rock when the algae are extremely thick instead of a 4 cm^2 scrape per rock. Remember to note this on the field sheets and

labels. If really thick, a diagonal section of the template can be scraped, but record the area.

6. Using a scalpel, completely scrape off the algae found inside the appropriate template.
7. The number of rocks and number of replicates collected will depend on the river and project - consult the project manager. For example, scrapes from three rocks have generally been combined on one filter, and three filters have been submitted per site. This should be pre-defined by the project manager prior to the sampling trip.
8. Place the algae from the scalpel directly onto a GF/C filter.
9. Apply a light sprinkling of powdered $MgCO_3$ to the material on the filter when all required rock scrapes for the replicate are finished.
10. Wrap the filter in the aluminum foil in a way that the analyst can easily unwrap it to get at the filter and so that material doesn't come off on the foil.
11. Label the wrapper with site, date, "epilithic chlorophyll", and the total area of scrape that it contains in cm^2 (e.g., three rocks x 4 cm^2=12 cm^2).
12. Repeat the process for the other groups of rocks.
13. Put the samples in a Whirlpak® or Ziploc® bag and store on regular or dry ice (-4 °C).
14. Place the samples in the lab freezer when you return from the field.
15. Ship frozen samples to the laboratory every week for extraction.

Method Variation

1. Add a small amount of deionised water to the freshly scraped algae on the rock to form a slurry and remove the slurry from the stone using a disposable pipette or a turkey baster, and transfer to a 250 ml dark Nalgene® bottle. Rinse the scalpel with deionised water in to the Nalgene® bottle to transfer any residual algae.

2. OPTIONAL: When all the rocks in a sample are scraped add 10-15 mg powdered $MgCO_3$ to the bottle.

3. Add double distilled/deionised water for an approximate total volume of 25 ml.

4. Label the bottles with the total area of scrape contained in the sample, site, date and "epilithic chloro".

5. Store the Nalgene® container at 4 °C and transport to the lab with in 24 hours.

6. At the lab, filter the bottle contents through a GF/C filter. Freeze or analyze the samples immediately.

Ash Free Dry Weight (AFDW)

1. Follow the rock sampling instructions described above for chlorophyll-a sampling (steps 1-4).
2. Consult with the project manager but consider taking a 2 or 4 cm^2 AFDW sample from the same stones sampled for chlorophyll-a. Thus algal scrapes for chlorophyll-a and AFDW would be taken from the same number and sub-set of stones. This facilitates integration of results and limits confounding factors but may only be possible with larger stones.
3. Use the 4 cm^2 template if small amount of biofilm is evident on rocks, or 2 cm^2 template if a large amount of thick algae is present.
4. The number of rocks and number of replicates collected will depend on the river and project - consult the project manager. For example, scrapes from three rocks could be

pooled into a small jar. This should be pre-defined by the project manager prior to the sampling trip.

5. Apply label denoting site, date, time, and total surface area scraped.
6. Put sample into cooler with ice and transport to lab within 24 hours. Store in deep freeze at lab until analysis

Species Identification

1. Follow the rock sampling instructions described above for chlorophyll-a sampling (steps 1-4).
2. Consult with the project manager but consider taking a 2 or 4 cm^2 Species Identification sample from the same stones sampled for chlorophyll-a and AFDW. Thus algal scrapes for chlorophyll-a, AFDW and Species Identification would be taken from the same number and sub-set of stones. This facilitates integration of results and limits confounding factors but may only be possible with larger stones.
3. The number of rocks and number of replicates collected will depend on the river and project - consult the project manager. For example, scrapes from two rocks could be pooled into scintillation vial containing 10 ml of deionised/RO water. This should be pre-defined by the project manager prior to the sampling trip.
4. Add 2 ml of Lugol's solution to each vial.
5. Line the scintillation vial cap with Parafilm® prior to sealing the vials.
6. Label each vial with site, location, date, area of scrape and sampler's initials.
7. Store vial in the dark.

Reference

This protocol was derived from Alberta Environment (2002), Biggs and Kilroy (2000), and BC-MWLAP (2003)

Collar Epilithic Method

Purpose

This technique was developed to address the concerns of the 'whole rock brushing' technique. It is to be used in the same circumstances as this old technique, i.e., clean looking but slimy rocks where the scalpel and template technique would not work, usually common in headwaters streams.

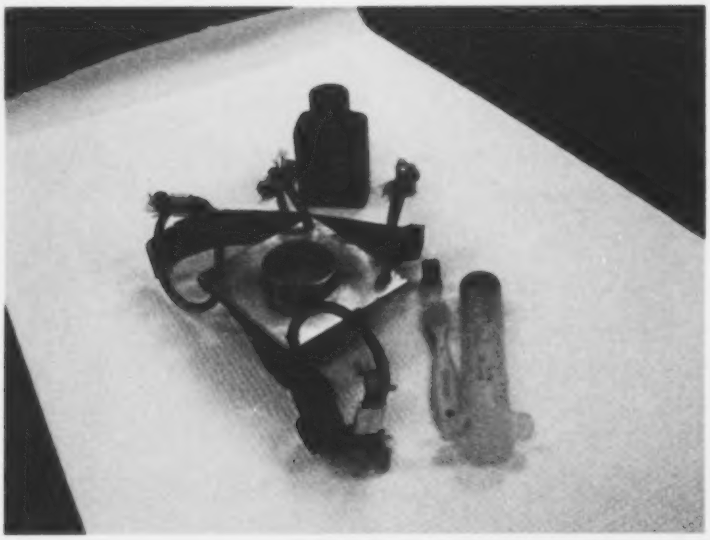

Figure 3.7-2 Epicollar

General

- To further standardise sampling effort, sample a sub-set of pre-defined conditions (depths of 40 cm, velocities of 0.3-0.5 m/s, only cobble sized substrates, etc.). This may be particularly useful when control sites are quite different to impact sites. Discuss with the project manager.
- Discuss the site selection and the collection and quality control procedures with the project manager before going into the field. Often shady areas of the river will be avoided.
- Sample in an area that will have flowing water covering the rocks throughout the open water season. Watch for changes of stage prior to sampling trip, as high stage might mean sampling recently exposed substrate.
- Ensure that the stiff brush is cleaned between scrapes.
- Keep the algae out of direct sunlight.
- Record full description of the site - flow, % algae cover, description of algae, recent changes in water level, etc. Take photographs of each site from upstream and downstream directions.
- Keep all acids, including that from skin, away from rocks, filters, and any apparatus used as this can degrade chlorophyll. $MgCO_3$ can be added to neutralize any acids present in the sample.
- Invertebrates must be excluded (the large ones, to a reasonable extent) since they eat algae. Avoid leaving any small stones on the filter as rocks will damage the laboratory high speed homogenizer used to grind the samples, and could endanger the laboratory technician.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Stainless steel collar with Neoprene® and bungee tie downs.
- Stiff artists paint brush
- Nalgene® filter apparatus with hand pump and pressure gauge
- Water squirt bottle, Nalgene® 150 ml dark bottles and labels
- Glass or plastic funnel (approx. 10 cm. diameter)
- Graduated cylinder, plastic syringe, turkey baster
- Labelling tape and waterproof markers and field sheet/book
- GPS unit, camera, tape measure.
- Disposable unpowdered latex, long rubber gloves.
- Safety equipment (see Section 4.4).

Procedure

1. Follow the rock sampling instructions described above for the template sampling technique for chlorophyll-a (steps 1-4).
2. The number of rocks and number of replicates collected will depend on the river and project - consult the project manager. This should be pre-defined by the project manager prior to the sampling trip.
3. Select a rock and affix the collar over an area of rock that was oriented upward in the stream.
4. Use artists brush to physically rub the area of rock within the collar area, to dislodge the slime.
5. Use a small amount of water and produce a slurry within the collar that can be transferred using a turkey baster or poured into a 1 litre Nalgene® dark bottle. If pouring use funnel to avoid spillage. Use squirt bottle to rinse slurry thoroughly from collar, baster and brush into the dark bottle. Try and use minimal water.
6. Repeat this process for three rocks ensuring enough slurry is obtained as required by project biologist. At this point, two directions can be taken.

    a. Shake some $MgCO_3$ into the bottle (OPTIONAL), store bottle (properly labelled with site, date, area sampled by collar X number of rocks) in cooler until such time as sample can be filtered. This should occur not more than 24 hours later.

    Immediately set up the filter apparatus with GF-C filter, rinse filter, and proceed to filter the slurry through the apparatus. Rinse bottle adequately and filter to ensure all slurry is obtained. Cover the filter with powdered $MgCO_3$ (OPTIONAL), fold the filter in quarters, place in aluminium foil, and label with date, location, site number, total area of rock sampled, and sampler's initials.

    b. If too much slurry is obtained, a sub sampling procedure may be used as follows:

        - Mix the slurry in a shallow graduated cylinder.
        - Draw up 10 ml of well mixed slurry into the syringe, 5 ml if the slurry is extremely thick.

  - Filter the slurry through a GF/C filter.
  - Rinse the syringe with a small amount of distilled water and filter this through the same filter.
  - Cover the filter with powdered $MgCO_3$ (OPTIONAL), fold the filter in quarters, place in aluminium foil, and label with date, location, site number, total volume of slurry (using graduated cylinder), volume of slurry filtered and sampler's initials.

c. Put samples on ice, or freeze with dry ice and deliver to the lab.

Reference

This protocol was derived from Biggs and Kilroy (2000), Alberta Environment (2002) and BC-MWLAP (2003).

Sand Coring Method

Purpose

The sand coring method is used to quantitatively sample epipsammic benthic algae for chlorophyll-a or biomass determination. Some watercourses or sections of watercourses have sandy or silty substrates which support communities of epipsammic algae. The template method is not appropriate for these soft substrates because algae are not attached to a defined surface, rather the algae are intermixed with the upper sediment layers. Coring techniques are far more effective to quantitatively sample algae from the upper layers of soft substrates.

General

- The depth of overlying water above the sediment, depth of each core, and number of cores sampled are to be decided by the project manager.
- Discuss the site selection and the collection and quality control procedures with the project manager before going into the field. Often shady areas of the river will be avoided.
- Keep the algae out of direct sunlight.
- Record full description of the site - flow, % algae cover, description of algae, recent changes in water level, etc. Take photographs of each site from upstream and downstream directions.
- Keep all acids, including that from skin, away from rocks, filters, and any apparatus used as this can degrade chlorophyll. $MgCO_3$ can be added to neutralize any acids present in the sample but this optional.
- Invertebrates must be excluded (the large ones, to a reasonable extent) since they eat algae. Avoid leaving any small stones on the filter as rocks will damage the laboratory high speed homogenizer used to grind the samples, and could endanger the laboratory technician.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Sediment corer (KB or 4-bore), Plexiglas® core liners and liner caps
- Core extruder and polypropylene filling funnel
- Glass filtering apparatus
- Nalgene® bottles (300 ml)
- 90% acetone and dispensette
- Magnesium carbonate solution
- Labelling tape and waterproof markers and field sheet/book
- GPS unit, camera, tape measure.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Safety equipment (see Section 4.4).

Procedure

1. Place a clean Plexiglas® liner in the barrel of the corer until the O-ring fits snugly.
2. Push the corer into the substrate and affix a rubber stopper to top of the tube.
3. Lift up the corer but before it breaks the water surface place a cap on the bottom of the liner.
4. Remove the liner from corer and place a cap on top of it.
5. Remove the bottom cap and quickly place the liner on the core extruder and remove top cap.
6. Carefully push the liner down; this will dispel the overlying water.
7. Place the core slicer on top of the liner and push the core up into the slicer until the required depth of core is obtained.
8. Slice off the core and transfer it to a glass filtering apparatus equipped with a GF/C filter.
9. Gently (7 psi or 48 kPa) vacuum the core until dry.
10. Add 2 ml of saturated $MgCO_3$ per core (OPTIONAL) and vacuum to remove water.
11. Carefully place the filter and core material in a 300 ml Nalgene® container.
12. Rinse (with acetone) any remaining material on the funnel into the Nalgene® container. Add approximately 25 ml of acetone for each core.
13. Shake the core/filter/acetone mixture for 1 min.
14. Label the Nalgene® container and field sheet with the date, site, depth of water, depth of core, number of cores, volume of acetone used and sampler's initials.
15. Cool to 4°C and transport to lab.

Reference

This protocol was derived from Biggs and Kilroy (2000), Alberta Environment (2002) and BC-MWLAP (2003).

3.8 Macrophyte Sampling

The aquatic macrophyte sampling methods detailed in Section 2.13 are generally applicable to river and stream sampling, particularly large river sampling. Alternative methods specific to rivers and streams are outlined in the following sections.

Routine Sampling - Large Rivers

Purpose

This method is qualitative or semi-quantitative. Purposes are as discussed in Section 2.13.

General

1. A PFD must be worn when wading in water. If depth and flow are high a U-Vic is recommended along with the use of a safety rope attached upstream of the sampling site.
2. Be careful when working amongst slippery rocks.
3. Wear gloves if there is a chance of faecal contamination in the water.
4. Only collect samples to a depth that can be safely reached.

Equipment

- sampling quadrat (e.g., 30.48 cm x 30.48 cm square)
- stakes for bench marks; sledge hammer
- measuring tape
- scissors or garden shears capable of cutting coarse vegetation
- plastic bags, 10-15 L capacity (or smaller if appropriate)
- waterproof markers
- waterproof notebook or field sheets and clipboard
- pencils and labels
- current meter & associated equipment
- depth stick
- light meter
- chest waders
- long gloves
- thermometer
- GPS unit
- U-Vics/PFD's
- throw rope
- digital camera

Procedure

1. The number of quadrats, transects, transect length, site selection and specific parameters measured depends on the particular study being done and should be discussed with the project manager prior to sampling.

2. At each location, establish a bench mark on shore to serve as a reference. Record UTM GPS coordinates for the bench mark. Use the same bench marks if surveys are to be repeated.
3. Measure out to the desired site or depth and record GPS coordinates. Take photographs to illustrate the sites and macrophyte growth.
4. Note the species present and relative abundance (%) of each; and growth robustness (Section 2.13).
5. Drop the quadrat in a random manner.
6. Remove all rooted plants from within the quadrat, allow plants to drain, and transfer to a labelled plastic bag.
7. The following measurements may be taken at each point as required:
   - depth
   - light readings - surface, sub-surface, mid point, bottom
   - velocity measured when >1 m depth @ 0.2 and @ 0.8 of depth and when <1 m @ 0.6 of depth
8. Record water temperature and all pertinent sampling information (date and time of sampling, GPS location, sample numbers, photo reference numbers).
9. When taking replicate samples, additional samples, or when returning to the same site, always move upstream to avoid sampling the same area twice in the same season.
10. If plant weights are required, follow the methods discussed in Section 2.13.
11. Samples to be archived or included in reference collections should be properly preserved in a plant press.

Reference

This protocol was modified from Alberta Environment (2002).

Peak Growth Sampling - Large Rivers

Purpose

This method is qualitative or semi-quantitative. Overall purposes are as discussed in Section 2.13. Specific objectives are to estimate peak standing crop.

General

General considerations are the same as for Routine Sampling.

Equipment

Equipment is the same as for Routine Sampling.

Procedure

1. Sample ten (10) random points at each site (5 right bank, 5 left bank) in the area of highest biomass (details may vary with study design).
2. At each marked sampling site, drop the quadrat in a random manner.

3. Note the species present and relative abundance (%) of each; and growth robustness. Example descriptors of robustness could include Dense, Moderate, Sparse and Trace. Take photographs to illustrate the sites and macrophyte growth.
4. Collect plants rooted within each quadrat in separate pre-labelled bags.
5. The following measurements may be taken as required at each point:
   - depth
   - light readings - surface, sub-surface, mid point, bottom
   - velocity measured when >1 m depth @ 0.2 and @ 0.8 of depth and when <1 m @ 0.6 of depth (method of determination dependent on depth)
6. Record water temperature and all pertinent sampling information (date and time of sampling, GPS location, sample numbers, photo reference numbers).
7. Do weight determinations as outlined in Section 2.13.

Reference

This protocol was modified from Alberta Environment (2002).

Transect Sampling - Small Rivers

Purpose

This method is quantitative. Purposes are as discussed in Section 2.13.

General

General considerations are the same as for Routine Sampling.

Equipment

- sampling quadrat (e.g., 1 m x 1 m square)
- 10-15 L plastic bags (5/site)
- waterproof markers
- waterproof notebook or field sheets and clipboard
- pencils and labels
- random number table
- 50 m measuring tape
- metre stick or wading rod (with cm graduations)
- plant identification keys [e.g., Brayshaw (1989); Burland (1989); Brayshaw (2000); Crow and Hellquist (2000a); Crow and Hellquist (2000b); Kershaw et al. (2001); and Lahring (2003)]
- chest waders, long gloves
- U-Vic's/PFD's, throw bag
- digital camera

Procedure

1. Select a representative 250 m reach of river.

2. Select 5 transects at 50 m intervals.
3. At each transect measure total width. Divide total width by 11 to obtain 10 sampling points.
4. At each sampling point, measure depth. Using the quadrat determine the first and second dominant substrates, macrophyte percent coverage and macrophyte algae percent coverage, according to the following:

Substrate Categories:	
Silt and Sand	<2 mm
Fine Gravel	2-16 mm
Coarse Gravel	16-64 mm
Cobbles	64-256 mm
Boulders	>256 mm

Percent Coverage Categories:	
Absent	0%
Sparse	1-30%
Moderate	30-60%
Dense	60-100%

5. Determine relative cover for species across transect.
6. Using a random number table, choose two sampling points at each transect, for macrophyte collection.
7. Collect all macrophytes using a quadrat as described previously.
8. If no macrophytes are present within the quadrat, record 0 in field notes, and select a third random number for macrophyte collection.
9. If $CaCO_3$ is present, remove with 5% acetic acid prior to spinning (optional).
10. Spin macrophytes 1 minute, or until all surface water has been removed.
11. Determine weights as discussed in Section 2.13.

Reference

This protocol was modified from Alberta Environment (2002).

3.9 Open Water Benthic Invertebrate Sampling

Purpose

Open water benthic invertebrate sampling involves the collection of invertebrates that inhabit the upper sediment layers and the sediment surface. Typically macroinvertebrates are sampled with some meiofauna, including early insect instars. The mesh size used to collect/process the invertebrate sample determines the composition of the benthic invertebrate sample collected. In streams and rivers, benthic invertebrates are collected from either erosional or depositional substrates and a variety of samplers are used (see project manager for preferred type of sampler). Open water benthic invertebrate sampling programs are usually conducted in early spring or fall, when benthic communities tend to be the most stable and relatively low flows facilitate sampling. It is also important to maintain consistency of time of sample collection within and between years.

There are a number of qualitative and quantitative sampling techniques each with advantages and disadvantages. The sampling program objectives and design will determine whether sampling should be qualitative or quantitative and the most appropriate sampler(s). However, the majority of benthic invertebrate monitoring programs tend to focus on quantitative sampling and so that will be discussed here for both erosional and depositional substrates.

General

- Verify the intent of the sampling program with the project manager. For many sampling designs it is necessary to standardize sampling locations with respect to habitat types. (i.e., choose specific sampling locations at each site so that similarities in substrate type, sampling depth and flow velocity among sites are maximized).
- Do not walk or stand in front of the sampler while sampling.
- Do not sample in eddies or backwaters.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Meter stick or depth sounder.
- Current meter & rod with earphones & batteries.
- Neill or Hess cylinder complete with net (212 μm mesh aperture).
- Labelled Nalgene® bottles, small shovel.
- Shoulder length rubber gloves and chest waders.
- Buffered Formalin.
- Waterproof field sheets/book and pens.
- Labelling tape and waterproof markers and field sheet/book.
- GPS unit, camera, tape measure.
- Disposable unpowdered latex gloves.
- Safety equipment (see Section 4.4).

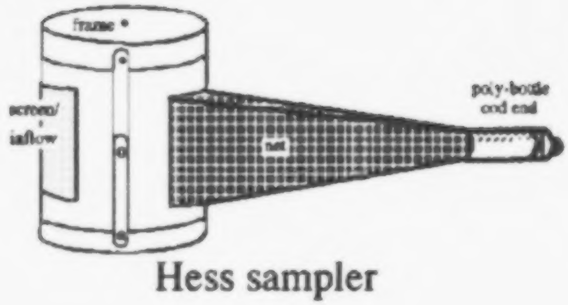

Figure 3.9-1 **Hess cylinder sampler**

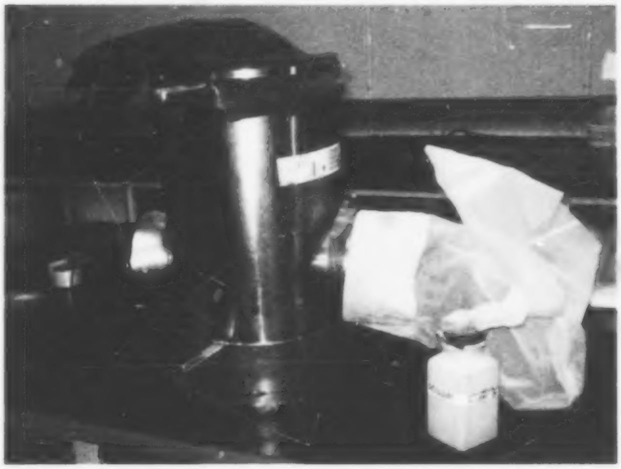

Figure 3.9-2 **Modified Neill cylinder sampler (Alberta Environment)**

Procedure

Erosional Substrates

The Neill or Hess cylinder sampler is one of the most commonly used benthic invertebrate samplers to sample erosional substrates in streams and rivers. This sampler is suited to a range of erosional substrate types such as gravel, cobble, small boulders and sand. Although, it is limited to a relatively shallow sampling depth in flowing water, a modified version of the Neill cylinder (mesh: 210 µm; substrate contact area: 0.1 m^2) has been used by the Alberta Environment to sample benthic invertebrates in major provincial rivers. Thus, in the interest of consistency Anderson (1990) recommended that this modified sampler be used to sample erosional habitats in Alberta Rivers. As a result, the Regional Aquatic Monitoring Program (RAMP) operating in the Athabasca Oil Sands Region, also uses a Hess or Neill-Hess cylinder sampler to sample erosional substrates (RAMP 2005). The following is a sampling procedure for operating the Neill or Hess cylinder sampler.

Neill or Hess Cylinder Sampler

1. Evaluate the study area to determine the dominant substrate type(s). Ensure this substrate is sampled at each site, and choose sites where there is sufficient current to inflate the sampler net.
2. Sample in depths of 30 to 50 cm of water.
3. Collect five samples per site, either in a transect perpendicular to shore or at random.
4. Label five Nalgene® bottles with site, date, location, sample number and sampler's initials.
5. Rinse net thoroughly between sample sites.
6. Ensure that the net is securely mounted on the Neill cylinder.
7. Screw a Nalgene® bottle onto the net receptacle.
8. Place the loose end of the net with the bottle attached, into the top of the cylinder.
9. Moving in an upstream direction, select an area of undisturbed substrate to sample.
10. Press the sampler into the substrate with the opening opposite the net facing the current. Feel inside the cylinder to ensure that there is a good seal. The teeth of the cylinder should be completely buried in the substrate.
11. If the seal is inadequate, rinse the net and bottle clean and select another sampling spot.
12. Once the cylinder is firmly anchored in the substrate, hold it there by standing on the lower handles.
13. Flip the net end with the bottle out of the cylinder and into the water.
14. Remove any large stones from inside the cylinder. Scrub them gently by hand and rinse them in the cylinder until no invertebrates remain attached to them. OPTIONAL: if not doing a visual characterization of substrate save these rocks to characterize substrate size.
15. Using the small shovel, stir the substrate for about 1 min. Ensure that the net does not clog as this will keep invertebrates from collecting in it. Gently stroking and shaking it will keep this from occurring.
16. Ensure that no particles escape out of the upstream opening of the cylinder. Stirring the sediment too vigorously, slow flows or the net clogging can cause this problem.
17. Let the inflowing water wash all the suspended particles from the cylinder into the net. The water in the cylinder should become as clear as the river water.
18. Gently stroke the net with your hands so that the particles in it move towards and into the Nalgene® bottle.
19. Lift the cylinder out of the water and repeatedly rinse the net by plunging it in and out of the water. Wash all particles and invertebrates into the Nalgene® bottle. Check for any invertebrates caught in the net and make sure they are collected.
20. Press the netting against the bottle mouth, invert the bottle and pour out most of the water. Turn the bottle right side up and splash the net with water to return any particles clinging to it into the bottle.
21. Unscrew the Nalgene® bottle from the net and preserve the sample with buffered Formalin immediately after completing the collections. FORMALIN IS A SUSPECTED CARCINOGEN SO HANDLE IT WITH EXTREME CARE. READ THE MSDS. Add approximately 1 part of full strength buffered Formalin to 10 parts of sample (if the sample contains a large amount of organic matter, algae and invertebrates, add approximately 1/5 the sample volume of buffered Formalin).
22. Determine depth of water at each sample location using a meter stick or calibrated shovel.

23. Use a current meter to obtain water velocity at 0.6 of total depth from surface, at each sample location. Count the number of revolutions in 60 sec. Use earphones if water is turbid.

Repeat the procedure for the remaining samples. Always sample upstream, and away from disturbed areas.

Take photographs of the site looking both upstream and downstream. Record and collect the following supporting data to characterise the benthic habitat at that site (see Appendix F for an example of a benthic invertebrate field data sheet).

- Water Depth: use a depth sounder, meter stick or velocity meter rod to measure the water depth at the approximate location that the benthic invertebrate sample was collected.
- Substrate Characterization: it is standard practice to characterise the sediment grain size of erosional substrates by visually estimating the percent aerial coverage of standard particle size categories according to classification systems (e.g., modified Wentworth classification system; the classification system given in Section 3.8). Particle size categories range from clay or silt to boulder and bedrock.
- Other: wetted and bankfull channel widths; GPS coordinates and site description; % macrophyte cover or qualitative description of epilithic algal cover; qualitative description of the amount of silt present.

Depending on study design, supporting information may also include.

- *In situ* Water Quality: Measure pH, DO, temperature and conductivity directly upstream of the approximate location that the benthic invertebrate sample was collected, according to the multiprobe methods described in Section 2.1. Measure flow using a velocity meter.
- Benthic Algal Communities: periphyton biomass (chlorophyll-a and/or AFDW; see Section 3.7)

Method Variation for Substrate Characterization

If not doing a visual substrate characterization (see above), use the rocks collected from inside the Hess sampler to characterize substrate size according to the following method.

Equipment

- Weigh scale and pan
- Tyler sieves #7 and #10
- 5 gallon pail
- Camera, metre stick, water proof field sheet/book and pens

Procedure

1. As each benthic cylinder sample is completed, remove all boulders, cobble, pebbles, sands and silts from the bottom of the cylinder before it is moved. Only remove material to the depth the shovel penetrated.
2. Place all material in the 5 gallon pail.
3. Transfer all material to the weighing pan and record total weight.
4. Remove boulder size material (>25 cm diameter). Weigh this material and record weight.
5. Remove cobble fraction (6.4 cm to 25 cm diameter). Weigh and record.
6. Pour rest of material through Tyler sieves: #7 (280 mm) on top of #10 (200 mm). Agitate sieves.
7. Weigh the coarse pebble fraction retained in sieve #7. Record weight.
8. Weight the fine pebble fraction retained in sieve #10. Record weight.
9. Subtract all the weights from the total weight to calculate the weight of the sand/silt fraction that passed through sieve #10. Record.
10. Place the four fractions from all site replicates side by side and take a photograph. Include a card with site name and date, and a meter stick in the photo.

Depositional Substrates

The procedure for sampling benthic invertebrate communities in depositional area of rivers and streams is similar to that described for sediment sampling in lakes. The type of grab sampler used will depend on sediment conditions and study objectives. Ekman dredge samplers are the most commonly used gear in shallow stream with predominantly depositional substrates.

1. Label the sample containers with site identification, sample type, sampling method, sampler ID and the date of collection. Take a photograph of the site looking upstream and downstream.
2. Note the following site/sampling information in the field sheet/book during the sampling process: target and actual sampling location (GPS); date and time of sample collection; overlying water depth (m); ambient weather conditions; grab penetration depth, depth sampled, sampling personnel; any deviations from the field sampling procedure.
3. Ensure that the dredge jaws open and close properly.
4. Lock the dredge jaws in the open position and lower in a controlled fashion to the stream bottom. Do not allow the sampler to "free fall". In streams with significant current, the dredge may be equipped with additional weight to prevent drifting on the decent and to facilitate vertical entry into the substrate. Deploy a messenger in deep water (>2 m) or use an attached pole or hand to trip the jaw mechanism in shallow water.
5. Slowly raise/lift the sampler off the bottom to prevent loss of fine sediment and then raise the dredge to the water surface.
6. The sample is deemed acceptable if the desired depth of penetration has been achieved; and the sampler has completely closed and was not inserted on an angle or tilted upon retrieval. If the sample does not meet these criteria sampling should be repeated close to the original sampling location. The rejected sample should be discarded in such a way that it will not affect subsequent sampling efforts.
7. Place a container (bucket or pan) beneath the sampler just as it breaks the water surface.

8. Open the grab sampler over a 250 or 210 µm mesh sieve box. Gently wash the sample using the sieve box to release the fine sediment and transfer the contents retained on the sieve to pre-labelled 1 L plastic jar(s). Use more than one jar if the sample is large. If more than one jar is used this should be recorded in the field book and indicated on each jar label (e.g., jar 1 of 3).
9. If on-site sieving is ineffective, be prepared to bag whole samples (i.e., double plastic bags) and provide cool storage and timely transportation to a lab.
10. Add buffered Formalin to the sample(s) to achieve a final concentration of 10%. See Appendix C for a protocol for the preparation of buffered Formalin. If the sample contains a large amount of organic matter, algae and invertebrates, add approximately 1/5 the sample volume of buffered Formalin.
11. Add a waterproof label with the sample ID to each sample jar (in addition to an external label) and securely cap the jar(s). Agitate the jar(s) to ensure the Formalin is evenly distributed throughout the sample(s).
12. Rinse the grab sampler and the sieve in stream water to thoroughly remove residual sediment, invertebrates or plant material.
13. In addition to a photograph of the site, supporting data must also be collected to characterise the benthic habitat at that site.

    1. Water Depth: use a depth sounder, meter stick or velocity meter rod to measure the water depth at the approximate location that the benthic invertebrate sample was collected.

    2. *In situ* Water Quality: Measure pH, DO, temperature and conductivity directly upstream of the approximate location that the benthic invertebrate sample was collected, according to the multiprobe methods described in Section 2.1.

    3. Substrate Characterization: Collect an additional grab sample at each benthic invertebrate sampling location within the lake. If the sediment sample contains a lot of excess water, place the sample in a pail and carefully decant off as much water as possible without compromising the silt and sediment content. Stir (homogenize) the sediment for 30 sec, then transfer into the appropriate pre-labelled containers for subsequent particle size analysis (grain size) and total organic carbon content. Store and transport samples in a closed cooler samples at 4 °C and do not allow to freeze. Make notes on colour and odour of sediment.

    4. Macrophyte cover: Where applicable, estimate % macrophyte cover at the sampling location and if possible list dominant species

Reference

This protocol was derived from Alberta Environment (2002) and RAMP (2005).

3.10 Under-Ice Benthic Invertebrate Collection

Purpose

In general, benthic invertebrates are collected in open water, in spring, late summer or fall. Still, in order to address specific issues such as the effects of low winter DO levels, it is sometimes necessary to sample benthic invertebrates under ice-covered rivers. Winter sampling is as late as ice conditions permit, usually March, to take advantage of warmer working conditions and longer days.

General

Follow the usual guidelines for open water sampling with the following additional equipment and procedures.

- This type of survey is best carried out with two crews - three people surveying and excavating openings in the ice and three people collecting the samples.
- Biofilm for AFDW and/or epilithic algae chlorophyll or taxonomic samples are often collected at the same time as benthic samples. Check with the project biologist for details and see Section 3.7 for sampling procedures.
- Ambient temperature should be between +5 and -15 °C to safely carry out this type of work.
- This is hazardous work - use extreme caution during all phases of sampling, and have the means to communicate with authorities in emergency situations. Follow all Safe Operating Procedures for ice cover work. See Section 4.4 for an overview of safety considerations and consult current Alberta Environment safety procedures for ice cover work.
- Sampling should proceed in a downstream to upstream direction to minimize disturbance to upstream areas yet to be sampled.
- Collect samples at sites with similar physical features (e.g., gravel/cobble substrates, moderate water depths, moderate water velocities).

Equipment

- Neill cylinder (Figure 3.9-2) with nylon bag fastened to top with a drawstring closure (0.1 m^2 sampling area; 212 μm mesh size)
- 2 SCUBA dry suits
- Ice augers and spud bars
- Chain saws (long blades)
- Ice picks and tongs
- Safety ropes and belts
- AFDW biofilm sampling equipment
- Current meter, GPS, metre stick
- Crew of six people
- Two skidoos with skimmers
- Equipment to process benthic invertebrate samples
- Equipment to process biofilm samples (see Section 3.7)

- ice fishing tent with a propane tank space heater

Procedure

1. Use ice auger to survey for possible sites, keeping depths, substrate, and flows as similar as possible between sites. Choice of a site may require deeper water than cylinder height; the nylon bag over the cylinder will prevent escape of invertebrates. Water up to 1 m deep may be sampled, depending on flow.
2. Use chain saws/augers to excavate a hole in the ice approximately 1 - 1.5 m wide and 2.5 - 3 m long, oriented with long part of hole in direction of flow. Use ice tongs to extract ice blocks, ensuring that substrate is undisturbed. A crew of three is needed to search for and excavate sites for sampling.
3. Attach guy ropes to bottom handles of cylinder, and safety ropes to the two persons in dry suits. Other ends of ropes should be fastened to ice picks pounded into ice for safety. Put shovel through top of cylinder bag and pull drawstring tight around handle.
4. During ice-removal and entry into the hole care should be taken not to disturb the benthic area to be sampled.
5. With one person on ice (upstream of hole) handling the cylinder guy ropes and safety ropes, the samplers enter the downstream end of hole and drill the cylinder into undisturbed substrate. Note that three people are needed to sample efficiently and safely.
6. The cylinder should be drilled into substrate far enough to ensure a good seal. Note that two people are needed in the water to ensure enough downward force to keep the cylinder anchored into substrate. Use the shovel to agitate the substrate at bottom of cylinder for approximately 2 min. Allow the cylinder to sit for 2-3 min to allow invertebrates to drift into net and bottle. Stroke net to prevent clogging.
7. Haul cylinder out of water and process sample following open water procedures described in Section 3.9, and put a new bottle onto net. The other person in water will do a velocity measurement with current meter at the sample location, as well as a depth measurement. Often, biofilm samples are required in triplicate from each hole, so nine rocks need to be collected. The person on ice can process the samples (Section 3.7).
8. Take necessary precautions to ensure that samples are not frozen during handling and storage. Samples can be stored in an insulated container (e.g., cooler) equipped with hot water bottles.
9. Take a photograph at each site and record supporting information such as: GPS coordinates, water velocity and depth, ice depth and substrate characteristics (visual assessment, see Section 3.9)
10. Proceed upstream of first replicate location about one pace, to obtain undisturbed substrate, and repeat the sampling procedure. The opening should allow collection of five replicates.

Reference

This protocol was derived from Alberta Environment (2002) and Golder Associates (2004).

3.11 Sampling Zoobenthos for Tissue

Purpose

The sampling of benthic invertebrates for tissue contaminant analysis is useful because it provides an indication of exposure or the bioavailability of contaminants in the sediment/water to biota. Bioavailability is a general term for the accumulation of substances in an organism or part of an organism, from water, substrate or food. This is important because not all contaminants in the aquatic environment are available for uptake by aquatic biota. For example, metals persist in the aquatic environment in many different forms depending on environmental conditions (metal partitioning in the aquatic environment). Some forms are more bioavailable, or available for uptake by biota, than others. The sampling of contaminants in benthic invertebrates provides a measure of the current status of contaminants in the aquatic system. Yet it is important to note that this does not demonstrate that the contaminant is causing a detrimental effect to the organism, it is simply a measure of exposure.

Specifically, the determination of contaminant bioaccumulation in benthic invertebrates identifies the presence and concentration of contaminants in whole organisms, or specific organs and tissues, depending on the study objective and logistics. Contaminants to be investigated will depend on issues and concerns specific to study objectives, but common examples include trace metals, polycyclic aromatic hydrocarbons (PAHs), pesticides and pharmaceuticals.

General

- The protocol given below recommends the use of Nitex® nets for the collection of benthic invertebrates. However, metal bioaccumulation studies conducted in the US have used kick nets (e.g., Cain and Luoma 1998; Maret et al. 2003; Caine et al. 2004). Nitex® is advantageous for sampling invertebrates for trace metal analysis because it can be acid-washed.
- Samples can be collected at discrete sites or throughout a particular reach depending on the objectives and design of the study.
- The different invertebrate taxonomic groups (Order, Family, or Genus) selected as Target Invertebrates for tissue analysis should be large enough and/or present in sufficient numbers to provide enough tissue for analysis. They should also be relatively easy to collect.
- The different invertebrate taxonomic groups (Order, Family, or Genus) selected as Target Invertebrates for tissue analysis should be widespread enough to enable comparisons among areas/sites.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Nitex® nets (of various meshes) mounted onto handles or Nitex® screens (of various meshes) mounted onto 2x4 frames
- Waders, long handled gloves
- Forceps (Teflon® wrapped or coated when the object is metal concentrations in tissue)

- Plastic Ziploc® bags/jars (for storing metals samples), glass jars (for storing trace organics samples) and labelling materials
- Polyethylene plastic (metals) or stainless steel pans (organics) – previously cleaned to either trace metal or trace organic standards (see Section 4.3).
- Unpowdered latex or polyethylene disposable gloves
- Waterproof field sheets/book and pens.
- Labelling tape and waterproof markers and field sheet/book.
- GPS unit, camera, tape measure.
- Safety equipment (see Section 4.4).

Procedure

1. Have one person hold the screen/net into the current of stream and another person disturb the upstream substrate with feet and/or hand.
2. Bring nets/screens to shore. Place the net contents into several pans for easier sorting of invertebrates; screens can be examined directly.
3. Pick out the target invertebrates for tissue analysis. Candidates for tissue analysis will be determined by the project manager and will depend on study objectives and design.
4. Invertebrates of different taxonomic groups (Order, Family, or Genus) can either be composited into one container, or into separate containers using forceps. Rinse the invertebrates with stream water (passed through the sampling mesh) prior to putting them into the sample containers to remove attached debris/sediment.
5. Sample container should be labelled with site name, date, taxonomic group and analysis required.
6. Samples should be stored on dry ice immediately and deep frozen to -70°C within 24 hours.
7. Adequate notes should be kept describing site conditions, substrate, physical variables (temp, pH, DO, conductivity), and site latitude/longitude.
8. Variations of this protocol may incorporate gut depuration/clearance procedures and procedures to remove contaminants adsorbed to the surface of the sampled invertebrates. Whether these variations apply should be decided by the project manager and depends on the objectives of the study.

Reference

This protocol was derived from Cain and Luoma (1998, Reynoldson and Rodriguez (1999), Alberta Environment (2002), Maret et al. (2003); and Cain et al. (2004).

3.12 Composite Sediment Grab Sampling

Purpose

In general, surficial sediment samples taken to assess recent sediment quality in streams and rivers are collected from depositional areas by Ponar or Ekman grabs. In lotic systems, sediments undergo frequent mixing and redistribution depending on the strength of water flow and frequency of high flows/flood events. Therefore, sediment quality can only be assessed in depositional habitats within watercourses.

General

- Samples should be taken from sites progressing in order, from the least contaminated site first to the most contaminated site last (the degree of contamination can be estimated from historical data, site conditions, land use, professional knowledge etc.).
- At any particular site, water sampling either should be conducted prior to sediment sampling to avoid disturbance of overlying waters by the sediment sampling technique or conducted directly upstream of the sediment sampling location.
- If the jaws are not closed properly when the dredge is retrieved, discard the sample and re-sample.
- See Section 4.3 for sampling equipment cleaning and decontamination procedures.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Bottom sampler – Ponar or Ekman grab sampler
- Rope (non-twisting nylon)
- Stainless steel or glass bowl or bucket
- Stainless steel or Teflon®/plastic spoons and slicers
- Labelling tape, waterproof pen, note pad, aluminium foil
- Hydrographic map showing sample sites
- Leak-proof sediment sample containers (e.g., widemouth glass jars, Teflon® lined lids)
- Lab analysis request sheets and chain of custody forms (COCs)
- Ziploc® bags, for samples, coolers and ice packs or hot water bottles, depending on the season.
- GPS unit, depth sounder, tape measure, camera.
- Pack of disposable syringes or turkey basters.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Equipment for field cleaning/decontamination procedures (see Section 4.3)
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals

Procedure

1. Ensure all equipment is cleaned to trace organic standards (see Section 4.3).
2. Discuss site selection and sediment requirements with project manager.

3. Label the sample containers with site identification, sample type, sampling method, sampler ID and the date of collection. Take a photograph(s) of the site/reach (upstream and downstream).
4. Note the following site/sampling information in the field sheet/book during the sampling process: target and actual sampling location (GPS); date and time of sample collection; overlying water depth (m); ambient weather conditions; grab penetration depth, depth sampled, sampling personnel; any deviations from the field sampling procedure (see Appendix F for an example of a benthic invertebrate field data sheet).
5. Ensure that the dredge jaws open and close properly.
6. Lock the dredge jaws in the open position and lower in a controlled fashion to the lake bottom. Do not allow the sampler to "free fall" The sampler should be in contact with the substrate or positioned just above it. Deploy a messenger in deep water (>2 m) or use an attached pole or hand to trip the jaw mechanism.
7. Slowly raise/lift the sampler off the bottom to prevent loss of fine sediment and then raise the dredge to the water surface.
8. The sample is deemed acceptable if the desired depth of penetration has been achieved; and the sampler has completely closed and was not inserted on an angle or tilted upon retrieval. If the sample does not meet these criteria the sample should be retaken close to the original sampling location. The rejected sample should be discarded in such a way that it will not affect subsequent sampling efforts.
9. The actual achievable penetration depth depends on the nature of the sediment and the sampling device used. A minimum penetration depth of 6-8 cm is recommended for surficial sediment samples but the preferred depth is 10-15 cm. These depths ensure minimal disturbance to the upper 2-5 cm of sediment that will be removed from the grab sample and submitted for physicochemical analysis.
10. Note the following sediment measurements/observations (where applicable): grab penetration depth; depth sub-sampled; type of material (soil type, colour, moisture condition, density, and grain size), biological structure (e.g. shells, large tubes, biota, macrophytes); debris (e.g., wood chips, plant or other fibres); obvious signs of anoxia (e.g., black layers); degree of sample disturbance; obvious odour or oily sheen; other unusual properties.
11. Siphon off any water on the surface of the grab sample with a syringe but if the water is cloudy allow to settle first (use a new syringe for every site). Remove the upper 2-5 cm of sediment with a stainless steel or Teflon® implement and transfer to a stainless steel tray/bowl. Avoid sediment at the edges of the grab sample (touching the grab sampler).
12. If more sediment is required to obtain the required volume for analysis, then more grab samples should be taken from the same site in undisturbed sediment. The volume of sediment to be submitted to the analytical laboratory depends on the study objectives, the parameters to be analysed and the analytical laboratory. The composite sample tray/bowl should be covered while grab samples are being collected. The number of grab samples collected should be noted.
13. Wash the dredge off in the lake. Rinse bucket and ladles before and after each site with river water. If sampling for organics, do an acetone/hexane rinse, collecting the waste in a jar for transport back to lab. Aluminum foil used to cover the bucket must also be rinsed with acetone/hexane (see Section 4.3 for detailed decontamination procedures).
14. Once sufficient sediment is collected, stir (homogenize) the composite sample for 30 sec, then transfer into the appropriate pre-labelled containers with a stainless steel implement.

15. Store and transport samples in a closed cooler samples at 4 °C and do not allow to freeze unless intended to be stored frozen. Place each sample container in two Ziploc® plastic bags (double-bag) in case of leakage.
16. If samples are not to be frozen and to be stored at 4 °C until subsequent analysis, the sample containers should be filled to the rim with no headspace to reduce oxygen exposure. This is particularly important if volatile parameters are to be measured.
17. If samples are to be frozen then a headspace of approximately 10% of the jar volume should be left in glass containers, to accommodate expansion of the sample when frozen. Check with the analytical laboratory before sampling to confirm sample storage requirements for the analyses requested. Samples stored at 4 °C in the dark generally can only be stored for days or weeks before analysis except for particle size analysis (grain size) which can be stored for up to 6 months. Samples can be frozen and archived for longer periods of time but it should be noted that samples destined for particle size analysis should not be frozen. See Environment Canada (2004) and USEPA (2002) for further information regarding sample storage and archiving.

It may be impractical to utilize a grab sampler where a thin depositional layer overlies compacted sediments that are too hard for a grab to penetrate. In such areas, the following 'bucket and spoon' method may be adopted.

Equipment

- Stainless steel bucket or Nalgene® bucket
- Stainless steel ladle and strainer spoon or Teflon® coated spoon
- Aluminum foil
- Widemouth glass jars, Teflon® lined lids, specially cleaned, for samples
- Ziploc® bags, for samples
- Coolers
- Ice/Dry Ice

Procedure

1. Ensure all equipment is cleaned to trace organic standards (see Section 4.3).
2. Discuss site selection and sediment requirements with project manager.
3. Walk along a 50-100 m reach at the site, from downstream to upstream.
4. Using the ladle or strainer spoon, collect the top 5 cm of sediment in the deepest water possible and transfer to the bucket.
5. Once enough sediment is collected, stir the composite sample for 30 seconds, then transfer into the appropriate pre-labelled containers.
6. Keep samples cool/frozen until stored or sent for analysis.
7. Record site description, samples collected, descriptions of sediment, etc.

Reference

This protocol was derived from Ontario MOE (1996), Ohio EPA (2001), Alberta Environment (2002), USEPA (2002), Environment Canada (2004) and RAMP (2005).

3.13 Air Lift Sediment Sampling

Purpose

This procedure is used to sample sediments in deep, moderately slow rivers.

General

- SAFETY: BE CAREFUL WITH STORAGE AND HANDLING OF THE COMPRESSED GAS TANKS. THEY ARE HIGHLY PRESSURED AND THE VALVE, IF DAMAGED, CAN CAUSE THE TANK TO TURN INTO A PROJECTILE. WHEN TRANSPORTING THE TANKS, BY TRUCK OR BOAT, IMMOBILIZE THEM IN AN UPRIGHT POSITION.
- The length of the upright steel tube used on the sampler will depend on the depth of water you are sampling - the optimum is to have water 2/3 the depth of the sampler.
- Check before sampling what analyses are to be done, the amount of sediment required and the type of container to be used (see Section 3.12).
- When finished sampling, clean the buckets according to procedures outlined in Section 4.3.
- PRESSURIZED TANKS CAN NOT BE TRANSPORTED BY HELICOPTER.
- Discuss sample site characteristics with project manager, i.e. should samples be collected in active flow, or backwaters, etc.
- See Section 4.3 for sampling equipment cleaning and decontamination procedures.
- See Section 4.4 for an overview of safety considerations/requirements.

Equipment

- Stainless steel air-lift sampler complete with hoses, hose roller
- Collector cup, gas nozzles and extension tubes
- Pressure tanks filled with nitrogen gas or compressed air (size "M")
- Regulator to fit the tanks
- 23 L capacity plastic buckets complete with lids (5-6 per site)
- 80 µm mesh triangular nets
- 10" crescent wrench
- Label tape, water proof pens and filed sheets/book, aluminum foil
- Stainless steel or Teflon®/plastic spoons
- Hydrographic map showing sample sites
- Leak-proof sediment sample containers (e.g., widemouth glass jars, Teflon® lined lids)
- Lab analysis request sheets and chain of custody forms (COCs)
- Ziploc® bags, for samples, coolers and ice packs or hot water bottles, depending on the season.
- GPS unit, depth sounder, tape measure, camera.
- Disposable unpowdered latex or polyethylene gloves, long rubber gloves.
- Equipment for field cleaning/decontamination procedures (see Section 4.3)
- Safety equipment (see Section 4.4).
- Waste containers for used chemicals

Figure 3.13-1 **Air-lift sampler**

Procedure

1. Remove the protective cap from one tank just before setting up.
2. Carefully attach the regulator to the tank - be careful not to over tighten as the brass threads will easily strip.
3. Attach hoses and the air lift sampler to the tank.
4. With the regulator closed, carefully open the main tank valve fully, then turn the valve back one full turn.
5. Slowly open the regulator until the pressure in the second stage reaches 20-45 psi (140-310 kPa).
6. Place the inlet of the sampler on the substrate - create a good seal by moving it around until it feels stable and set into the substrate.
7. Deliver short pulses of compressed gas to the sampler.
8. Collect the thickest slurry in clean buckets (pre-filter it through an 80 Fm mesh held under the sampler outlet to limit particle size).
9. Collect 5-6 buckets in total.
10. Move the sampler frequently.
11. Clearly label the site on the buckets and ensure that their lids are tightly sealed.
12. When finished sampling turn off the main valve on the tank, bleed pressure out of the line and unhook the hoses. Carefully remove the regulator and replace the protective cap on the tank.
13. Record in field notes: number and size of buckets collected; exact sampling location; flow conditions; turbidity of the river; amount and a description of the sediment; abundance of attached algae and macrophytes; date and time.
14. Upon return to the laboratory, let the sediment containing buckets stand for 20-24 hours.
15. Siphon off the supernatant, saving this in clean jar(s) as it may be analyzed for the same parameters as the sediment.
16. Put the remaining sediment in clean glass jars.
17. Let the jars sit for 24 hours in a fridge.

18. Remove the supernatant from the top of the jar and save with the previously collected supernatant.
19. Collect a minimum of one 500 ml jar of sediment.
20. Clearly label the jar with site and date.
21. Freeze the sample.

Reference

This protocol was derived from Alberta Environment (2002).

3.14 Sediment Oxygen Demand (SOD)

Purpose

Sediment oxygen demand (SOD) is a measure of the oxygen consumed by biochemical decomposition of organic matter in stream or lake deposits. SOD can be used to evaluate the impact of a point source input to the water quality of a water body, or as a metric (input) for use in water quality models. SOD over a defined time period can be measured either in situ using a chamber method, or sediment cores can be taken for subsequent incubation and SOD measurement in a controlled environment. A number of procedures have been developed but they are often specific to individual studies. As such there are a limited number of SOD manuals or standard operating procedures (SOPs) publicly available.

Chamber Method Procedure

This method refers to the measurement of SOD using an in situ chamber. However, it should be noted that this may not be applicable to all sites because of issues like vandalism or accessibility.

General

- Choose representative substratum for the site. Substratum composition ideally should be cobble and gravel interspersed with fines. Substrate should not be so large as to not fit inside the chamber.
- Chambers should be placed in areas of flowing water.
- Ideal working depth (from ice surface to substrate) is between 50-70cm, but no greater than 90 cm.
- Ensure that sufficient distance between bottom of ice and top of velocity vanes exists to allow vanes to spin freely.

Equipment

- SOD field sheet
- SOD chambers
- Ice auger
- Chainsaw
- Ice tongs
- Ice chisels
- Spades
- T-posts
- Rope
- Sledgehammer
- Meter ruler
- Extra stoppers and wire
- Long armed rubber gloves
- Depth-profile measuring device
- Depth-profile measuring rod.

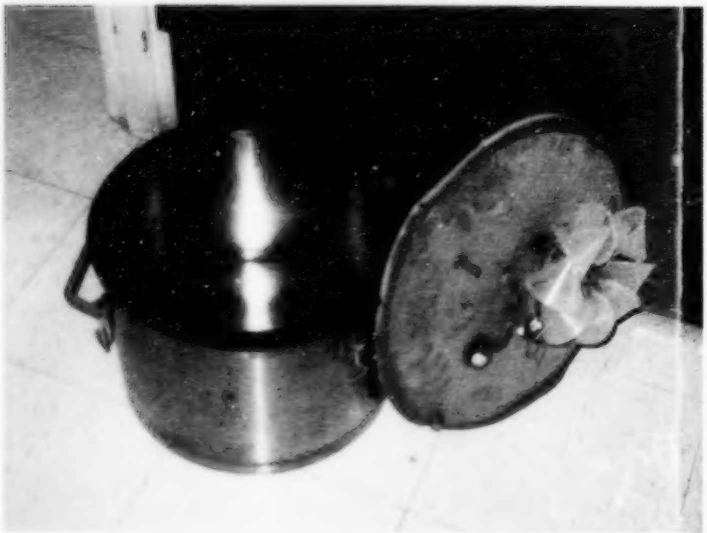

Figure 3.14-1 SOD chamber

Winkler DO equipment:

- Winkler bottles
- Tygon® tubing- 1/4"I.D. X 3/8"O.D.
- Winkler chemicals
- Winkler titration field kit

Other apparatus necessary for the measurement of other pertinent variables in the field, such as velocity meter, Hydrolab® Minisonde, epilithic algae field kit, etc.

Procedure

General Preparation of Chambers:

1. Check that each chamber has all stoppers attached to the lid (use light-duty chain and epoxy cement) and that rubber and foam gaskets are secured and in good condition on the chamber. Foam gaskets on ALL chambers should be completely changed before undertaking any new work
2. Ensure that the lid fits snugly on the chamber (each chamber has the same exclusive number for the lid and base) and that the snap-down clips operate correctly.
3. Check that the water vanes rotate freely and the Teflon® washers are in good condition.

Filling the Chambers:

1. A total of 4 chambers will be filled with substratum; and 1 "control" chamber to be filled with river water.
2. Choose representative substratum for the sites to be used in the study area.
3. Fill chambers with ambient water to about ¼ of its depth.

4. Fill ¼ of the chamber with finer material such as gravel and sand to form a base on which the larger cobble can rest on.
5. Using a shovel, carefully remove undisturbed cobble and gravel from the substrate and place atop the sand/gravel base in the chamber. Arrange rocks (epilithic cover must face up) such that they are representative of substratum conditions at the sample site.
6. Equal amounts and type of substratum should be placed in each chamber. Compare with other chambers.
7. The chambers are usually filled between 1/3 and 1/2 of the chamber depth, to leave room for the water vane unit on the inside of the chamber.
8. Place the chamber, without the lid, very carefully into the water and allow it to fill very gradually to reduce disturbance to the substratum in the chamber.
9. Position the chamber on the stream bottom in a level fashion.
10. Leave the chamber in position until the sediment in the chamber has settled or flushed. This is important since the suspended material in the water column may increase oxygen demand in the chamber.

Closing the Chambers:

1. Ensure that the rubber and foam gaskets and the water sample tube are free of ice or sediment.
2. Remove the stoppers from the portholes in the lid and place the lid gently onto the chamber, making sure that no bubbles are trapped between the flange and the lid.
3. In deeper depths, another chamber (that has been turned over onto the substrate) can be used as a platform to elevate the chamber to a suitable working depth.
4. Check that the foam gasket on the lid is snugly fitted onto the chamber base.
5. Ensure that the four snap-down clips on the chamber lid have been extended equally (clockwise to tighten, counter-clockwise to loosen) for easier clamping and to prevent warping of the lid.
6. Close 2 diagonally opposite (IMPORTANT) clips on the lid simultaneously, and repeat with remaining clips. It may be necessary to adjust the clips so that they snap down briskly onto the base, giving a secure fit.
7. Making sure that the chain is not wrapped around the base of the vane, replace the stoppers securely in the portholes. Having some slack in the chain will allow for some give if the chain is struck by ice or debris. IMPORTANT: Record time of lid closure for each chamber to the nearest minute on field sheet.
8. Depending on river velocity and site location, it may be necessary to anchor the chambers using rope secured to one or more t-posts pounded into the ice surface and tied to the chamber handles.
9. In higher velocity locations, rocks may be piled up against the base of the chambers for extra stability.
10. Measure river ambient D.O. concentrations with a Hydrolab® field unit and obtain 2 sets of Winkler samples for analysis upon return to the lab.

Obtaining Sample:

1. Take note of any unusual conditions: stopper not in porthole; retaining clip open; vane performance (spin or no spin); chamber tipped over, etc., and record on SOD field sheet
2. Rotate water vane of all the chambers very slowly to achieve complete mixing of water.

3. Remove from river and record chamber number and date and time.
4. Crack open the smallest stopper and while gently rotating the vane. Place Tygon® tubing over the water sample tube and carefully siphon water into the Winkler bottle. Note: Sample should be discarded if air bubble or ice is present in Winkler bottle. Replace Tygon® tubing if freezing occurs.
5. Preserve with Winkler chemicals.
6. Repeat steps 3-5 for all chambers.
7. After water samples for D.O. measurement have been taken, remove lid of the chamber.
8. In order to measure the volume of the chamber, place the depth-profile measuring device over the opening of the chamber and secure using the 3 alignment pins.
9. Place the depth-profile measuring rod (pre-marked in 0.5 cm increments) through one of the holes in the Plexiglas® plates until the rod touches the substratum. Record the depth as indicated on the rod and subtract 0.5 cm (width of the Plexiglas® as it sits above the chamber flange) from the total. This distance will give the distance from the top of the rock to the underside of the Plexiglas®. Record on field sheet.
10. Repeat for remaining 11 holes.
11. Record dissolved oxygen of the water within the chamber using a Hydrolab® Minisonde. This is used mainly as a check against the Winkler results.

Reference

This protocol was derived from Alberta Environment (2002).

4.0 GENERAL SAMPLING CONSIDERATIONS

This section of the manual deals with issues of importance to the success and integrity of a surface water quality sampling program. Specifically, an overview of QA/QC requirements, additional sampling measures for sampling trace organic compounds in surface waters, equipment cleaning procedures for surface water and sediment quality sampling, and an overview of safety considerations.

4.1 Overview of QA/QC requirements for Surface Water and Sediment Quality Sampling

Quality Assurance/Quality Control

The key to ensuring the samples submitted to the laboratory have not been subject to contamination or degradation is an effective QA/QC program designed to meet clear data quality objectives (DQOs). Environment Canada (2004) defines Quality Assurance (QA) as "a planned system of operations and procedures whose purpose it is to provide assurance to the client that defined standards of quality are being met." Quality control (QC) is a part of the overall quality assurance program and QC procedures/practices ensure that the quality of the data collected meet pre-established performance criteria and standards (i.e., DQOs). The samples should be submitted to an accredited laboratory for analysis.

Data Quality Objectives

Data quality objectives (DQOs) are qualitative or quantitative statements that clarify study objectives; define the appropriate type of data; and define the level of uncertainty that is considered to be acceptable in the data (USEPA 2000b). If the data do not meet the qualitative or quantitative criteria defined by the DQOs, then a series of corrective actions must be taken to ensure quality control and data integrity.

Quality Assurance/Quality Control Sampling Program

A QA/QC sampling program should include: the field quality assurance, quality control samples, and laboratory QA/QC procedures.

Field quality assurance includes basic precautions that must be followed if imprecision and bias in the data are to be minimized. Examples of quality assurance include:

- this field sampling manual, which provides specific instructions to maintain consistency and ensure the staff are diligent while collecting, preserving and shipping samples;
- the maintenance and calibration of all field equipment; and
- proper documentation of required information (USEPA 2000b; USGS 2005).

Quality control samples are used to evaluate whether the sampling and processing system is functioning properly, and whether the measurement quality objectives have been met. Analytical labs have their own quality control procedures, but QC samples submitted from the field will provide an estimation of the total study error. If necessary, QC samples can be used to pinpoint sources of error, especially those from contamination. New sampling programs should incorporate rigorous QC measures until an acceptable level of data quality has been demonstrated, especially if the program objectives are to investigate an impact on aquatic life or human health.

Surface Water Quality QA/QC Samples

The number of QC samples that should be collected relative to the total number of samples is presented in Table 4.1-1. This is a general guideline only that should be varied depending on the program objectives. For example, one would collect more QC samples when investigating a problem that affects aquatic life or human health (up to 35%), and fewer when routine samples are collected to monitor water bodies without major issues, such as recreational lakes (5 - 10%). In general, when the substances of concern are at trace levels, more blanks should be collected; if concentrations are high, more replicates should be collected. The following is from Government of British Columbia (1998), USGS (1997) and Alberta Environment (2002).

Table 4.1-1 General guidelines for types of quality control samples and their frequency of collection (excerpted from Mitchell 2006)

	*Frequency**	*Quality Control Checks*	*Comments*
Blanks			
Field	1/10	Contamination from bottles, collection methods, atmosphere, preservatives.	Treat as real sample.
Trip	1 per trip	Contamination from bottles	For volatile compounds only.
Equipment	1/Every fifth site	Carry-over between sites	When carry-over possible.
Filtration	As needed	Contamination from filters or filtration equipment	When contamination is suspected.
Bottle	As needed	Contamination from improper cleaning	Only for bottles cleaned by field staff.
Spikes	1/20	Bias	Only for variables of concern
Replicates or Splits	1/10	Lab and field precision	Only for variables known to be above detection limit
Reference	Once at beginning of season or new project	Accuracy	Performance test before sampling begins

Notes: * - 1/10 = 1 QC sample for every 10 regular samples

QA/QC samples should include at a minimum, the collection and analysis of field blanks, trip blanks and duplicate samples. QA/QC samples may also include: equipment blanks, triplicate samples, split samples and field spikes. The collection and analysis of these samples is designed to detect unintentional systematic and/or random errors in the actual sample data. If field and trip blanks are not collected this error may go undetected and so may be mistakenly attributed to the analytical laboratory.

Blanks: Blanks are de-ionized water carried into the field. They can identify unsuspected contaminants from the water itself, improper cleaning procedures, preservatives, samplers, filters, travel, sampling techniques or air contaminants. Field blanks are exposed to the sampling environment at the site and handled as a "real" sample. They provide information on contamination from all phases of sampling and analysis. They should be done routinely and at a reasonably high frequency. Other types of blanks are used to pinpoint problem areas. Bottle blanks are prepared sample bottles filled with de-ionized water and used to check for contamination from the bottles, their cleaning or preservatives. Trip blanks are bottles filled with carbon-free de-ionized water at the lab and transported from the field without being opened. They are generally for volatile compounds only. Equipment blanks are samples of de-ionized water that has been used to rinse sampling equipment. Filtration blanks are those that have passed through the filtration apparatus in the same manner as the sample. These can pinpoint cross-contamination through inadequate cleaning techniques. In general, blanks should be analyte-free, or at a minimum, 5 times lower than the concentration of any variable in the real sample.

Reference Samples: Reference samples are used to document the bias and precision of the analytical process. The simplest type of reference sample is provided by a laboratory that is not involved in analysis of the real sample. The reference sample is prepared by adding a known quantity of the variable of interest to a given amount of pure water. Another type of reference sample is the Certified Reference Sample (CRS). It is a portion of a very large batch sample that was collected from one place at one time. It has been analyzed by a large number of independent labs, and therefore the distributing agency can provide a mean and confidence interval for the variable of concern. It is obtained from a scientific body such as the National Research Council. Although laboratories use reference samples for their own QC, a reference sample should be submitted blind to the lab before each new sampling program begins. A submission of multiple samples of the same CRS yields the laboratory precision, as well as bias.

Spikes: Spiked samples are prepared by spiking aliquots of a single water sample with pre-measured amounts of a reference sample. An aliquot of the same sample is left unspiked. The difference in concentration of the two samples should equal the amount of the spike. This reveals systematic errors (bias) in the analytical method. Bias can arise from matrix interference or analyte degradation. Accuracy is estimated by calculating the amount of the analyte in the spiked sample and subtracting the amount in the unaltered duplicate sample. Recovery is the percentage of the recovered amount of analyte.

Replicates: Replicates are used to calculate precision. Two or more samples collected in quick succession from the water body will yield precision of the entire program. It also includes variability of the water body itself, but this should be minimal compared with other sources of variability. Replicate results that are non-detects cannot be used to estimate precision, so the project manager should select samples or measurements likely to yield positive results.

Splits: A split is a single sample that has been divided into two or more equal portions. If the sample is split in the lab, these can check laboratory precision. If done in the field, these yield field + lab precision. When splits are done in the lab and field (i.e., one of the field splits is split again in the lab), the lab precision can be subtracted from the total precision to yield field precision. Sometimes splits are sent to different laboratories for analysis. However, there is no way to determine which laboratory's results are correct. Instead, the project manager should submit reference materials or do a performance test before engaging the lab. Non-detect results cannot be used to assess precision.

Sediment Quality QA/QC Samples

Typically the total number of QA/QC samples should represent a minimum of 10% of the total number of sediment samples. They should include at a minimum, the collection and analysis of field duplicate samples and split samples. QA/QC samples may also include: equipment blanks, field blanks, triplicate samples and field spikes (USEPA 2001).

Laboratory Selection

Water and sediment quality samples should be submitted to laboratories accredited by the Canadian Association of Environmental Analytical Laboratories (CAEAL) and the Standards Council of Canada (SCC) as specified in the project design. Under this accreditation, laboratories are required to participate in an annual performance evaluation of procedures, methods and internal quality control. Specific internal quality control procedures are performed to guarantee analytical precision and sample integrity during sample analysis (e.g., spiked samples; duplicate samples; method blanks; reference standards).

4.2 Trace Organic Sampling Considerations for Rivers and Lakes

Trace Organic Water Sampling

Trace organic sampling in surface waters can be completed in combination with discrete or composite sampling for other parameters, as long as consideration is given to the following points, limitations and additional/replacement sampling techniques.

- Consult with project manager and analytical lab for the precise sample types and methods required.
- Only glass, Teflon® or stainless steel, cleaned to trace organic standards, should contact the sample.
- Water samples collected for trace organic analyses should not have any headspace and should be collected in amber of dark glass bottles to limit potential photo-degradation.
- Pour samples with minimum of aeration to minimize volatilization.
- Avoid using an intermediate container to fill sample bottles as it may cause contamination.
- AOX and PPCP must be preserved immediately in the field.
- *Sampling Under Ice:* Volatile Priority Pollutants (VPP) vials (Alberta Research Council Laboratory) are difficult to fill under ice - ensure the auger hole is well flushed and lower the vials by hand as deep as possible, then cap below the surface. Alternately, a clean trace organic 1L brown bottle can be filled and VPP vials immediately poured off from that, ensuring no head space.
- *Blanks:* Use ultra-pure deionised water supplied by the appropriate analytical lab section for field blanks. This should be the highest-grade deionised water used in the analysis of the trace organic parameters being sampled. water.

Trace Organic Sampling Procedures

These procedures are in addition to those described in Sections 2.4-2.6 and 3.3, and summarized in Table 4.2-1.

Equipment

- Trace organic bottles (specially cleaned)- supplied by analytical lab
- Stainless steel holder with foam insert

Procedure

1. Label bottles with the site name, date sampled, and sample type.
2. DO NOT RINSE BOTTLES and DO NOT TOUCH THE INSIDE OF THE LID OR BOTTLE MOUTH
3. Pesticide or trace organic sample bottles or vials are always glass, and should be filled directly from the sample medium, or by using a trace organically cleaned intermediate container, usually of stainless steel.

Table 4.2-1 Specific sampling procedures for sampling trace organic contaminants in surface waters

Surface Water	Sampling Conditions	Specific Sampling Techniques*
Rivers	Open Water	Submerse the bottle directly in the flow facing upstream. Cap below the surface when full
	Under Ice	Place bottle in stainless steel holder with foam insert - drop quickly into the main flow below the ice. Cap below the surface when full.
	Off Bridge	Place bottle in stainless holder with foam insert, keep cap of bottle on until bottle is inserted in place. This avoids the risk of touching the bottle mouth to the stainless holder. Lower into main flow, raise bottle up and down in the flow until full, then carefully raise to bridge level and cap
Lakes	Open Water or Under Ice	Use peristaltic pump to collect a vertically integrated comp - tubing should be Teflon®. Submit a QA/QC pump equipment blank to check for possible contamination

* These procedures are in addition to those described in Sections 2.4-2.6 and 3.3.

Surface Film Sampling

Purpose

The purpose of conducting this type of sampling is to determine the presence of trace organic contaminants in surface films present in surface waters.

General

1. Avoid coming in contact with dichloromethane, it may cause irreversible health effects. Ensure you have read the MSDS.
2. Ensure glass plate and Teflon® funnel are properly cleaned and stored before and after sampling.
3. Try to collect the sample using only 100 to 200 ml of DCM. Too much DCM hinders analytical recovery of the contaminants.

Equipment

- 1 L trace organic bottle – supplied by analytical lab
- Teflon® funnel
- 20 cm x 20 cm glass plate 5-6 mm thickness with a C-shaped handle attached to one side of glass plate
- Dichloromethane (DCM)
- Teflon® 500 ml squirt bottle
- Neoprene® or other suitable safety gloves

Procedure

Note: The following procedures are difficult under windy (wave) conditions.

1. Label bottles with the site name, sample date and sample type.
2. DO NOT RINSE BOTTLES.
3. DO NOT TOUCH THE INSIDE OF THE LID OR BOTTLE MOUTH.
4. Wearing suitable safety gloves, rinse the Teflon® funnel with DCM from the squirt bottle. Catch all waste DCM into a closed container.
5. Hold the glass plate by the handle and rinse the other side of plate using DCM from the squirt bottle. Clean entire area of glass plate and capture all waste DCM into a closed container.
6. Gently lower the glass plate until it comes in contact with the surface of the water. The plate and the water's surface will form a natural adhesion. Hold plate to the surface for a fraction of a second, then remove from water's surface. Be careful not to submerse the plate below water surface.
7. Insert Teflon® funnel into trace organic bottle.
8. Rinse glass plate with DCM squirt bottle into the Teflon® funnel. Rinse entire surface area of glass plate, collecting all the DCM into trace organic bottle.
9. Complete a composite sample by collecting samples at other locations of the water body.
10. Collect about 100 ml – 200 ml of DCM.

4.3 Cleaning Equipment Requirements for Surface Water and Sediment Quality Sampling

Purpose

The effective cleaning of sampling equipment prevents, minimizes or limits the cross-contamination of samples taken during a sampling trip or between sampling trips. This is particularly important when trace parameters such as trace metals, or trace organic constituents are being sampled. Cleaning is also used to remove manufacturing residues from new equipment, and to remove dust and any other foreign substances that has been in long-term storage or transport and requires cleaning.

Sampling equipment should be either disposable (single only use) or subjected to rigorous cleaning procedures (depending on the parameters being sampled) and stored in sealed new plastic bags (e.g., Ziploc®) or wrapped in new aluminium foil, depending on what parameters are to be sampled. Parafilm® (self-sealing, moldable and flexible film) can also be used to cover equipment openings to prevent dust accumulation.

All sampling bottles should be obtained from the appropriate analytical laboratory, only used once, remain capped before and after actual sampling, only be used for that specific sampling procedure, and not to be cleaned. The following cleaning procedures are for all non-disposable sampling equipment excluding sample bottles.

General

- A 5% (v/v) solution of Hydrochloric acid or a 10% (v/v) solution of Nitric acid should be used to soak/rinse equipment that is to be used to collect samples for trace metals analyses. Note: if the equipment is to be used to collect samples for nitrogen analyses then Nitric acid should not be used. ALWAYS ADD ACID TO WATER.
- A non-phosphate, laboratory-grade detergent (e.g. Liquinox®, Contrad®, Extran®) should be used to soap-wash equipment. Use a 0.1-2.0% v/v solution when cleaning between field trips (higher if required) and use a 0.1-0.2% v/v solution for field-cleaning. Do not use >0.2% v/v solution for field cleaning. To limit soap residue accumulation, do not use >0.2% v/v solution for field-cleaning
- Use a fume hood if one is available. If not, wear respirators with the appropriate filters for the hazardous chemicals in use (e.g., acids, solvents). Clean equipment in a well ventilated area, and wear safety glasses and gloves.
- Consult the MSDS sheets for all chemicals used in the cleaning procedures for information regarding personnel protection, spill clean up, and medical treatment directions.
- Note: all containers or equipment for trace organic work must be stainless steel, glass or Teflon®.
- Equipment or rinsate blanks can be collected to test whether there is any residual contamination left on the equipment after cleaning. A rinsate blank is a sample of deionised water collected after it has been poured over/through the sampling equipment

in question. The water should be the highest grade water used to analyse the samples collected by that piece of equipment.

- If methanol is used as a solvent then do not use it on equipment destined to be used to collect samples for total particulate carbon (TPC), particulate organic carbon (POC), or total or dissolved organic carbon (TOC/DOC).

Cleaning Equipment for Trace Inorganic Sampling

The recommended cleaning procedure for sampling equipment used to collect samples destined for the analysis of trace inorganic parameters (e.g., trace metals) is:

1. Physical removal with brushes and non-phosphate detergent wash. This removes all visible particulate matter and residual oils and grease.
2. Tap water rinse and distilled/deionised water rinse. This removes detergent residues.
3. Acid rinse or soak non-metallic sampling equipment. Use either a 5% (v/v) solution of Hydrochloric acid or a 10% (v/v) solution of Nitric acid. Typically the equipment is subject to either an acid rinse or soak between field trips, and an acid rinse when cleaning/decontaminating equipment in the field. Consult with the project manager regarding the procedure to be used.

   Acid Rinse: Equipment should be rinsed thoroughly with acid, ensuring that the acid makes contact with all surfaces likely to be in contact with the sample.

   Acid Soak: Ideally equipment should be soaked for 12-24 hours, but USGS (2005) recommends a 30 min acid soak. Consult with the project manager regarding the length of the acid soak.

Ideally both procedures should be conducted in a fume hood but if that is not available follow the safety instructions given in the Section 4.4.

4. Step 4: Multiple distilled/deionised water rinses (3-5 rinses). At a minimum the last rinse should be deionised water. To remove all acid residues ensure that the water makes contact with all surfaces likely to be in contact with the sample.
5. Step 5: Air dry in a clean area and on a clean non-metal surface. Avoid areas with dust and fumes.
6. Step 6: Store in new, clean Ziploc® plastic bags, and/or cover equipment openings with new Parafilm®. Mark the date of cleaning and the initials of the cleaning personnel on the bag. Store and transport the cleaned equipment in a clean vessel, in a clean area.
7. Step 7: Discard waste acid into a clearly marked waste jug for acids and store in hazardous waste area for proper disposal. Consult the MSDS sheets for details and do not store acids close to solvents.

Cleaning Equipment for Trace Organic Sampling

The recommended cleaning procedure for sampling equipment used to collect samples destined for the analysis of trace organic parameters (e.g., hydrocarbons) is:

1. Physical removal with brushes and non-phosphate detergent wash. This removes all visible particulate matter and residual oils and grease.
2. Tap water rinse and distilled/deionised water rinse. This removes detergent residues.
3. Rinse with organic solvents (e.g., acetone, hexane, methanol). A common procedure is to first rinse with hexane and allow to air dry and then rinse again with acetone and allow to air dry. Equipment should be rinsed thoroughly with the solvent, ensuring that the solvent makes contact with all surfaces likely to be in contact with the sample.

Ideally both procedures should be conducted in a fume hood but if that is not available follow the safety instructions given in the Section 4.4.

4. Multiple distilled/deionised water rinses (3-5 rinses). At a minimum the last rinse should be deionised water. To remove all solvent residues ensure that the water makes contact with all surfaces likely to be in contact with the sample.
5. Air dry in a clean area and on a clean surface, cover the surface with new, clean (rinsed in hexane/acetone) aluminium foil. Avoid areas with dust and fumes.
6. Cover the equipment with new, clean aluminium foil and store in new, clean Ziploc® plastic bags. Mark the date of cleaning and the initials of the cleaning personnel on the bag. Store and transport the cleaned equipment in a clean vessel, in a clean area.
7. Discard waste hexane/acetone into a clearly marked waste jug for organic solvents and store in hazardous waste area for proper disposal. Consult the MSDS sheets for details and do not store acids close to solvents.

Cleaning Procedures for Specific Pieces of Equipment

Euphotic Sampling Tube/Peristaltic Pump Tube

(Tygon® type R 3603 size 3/8" x 1/8" and 1/2" x 1/8")

1. After use, rinse with deionised/distilled or tap water inside and out.
2. Remove foot valve from euphotic tube and soak in 5% HCl.
3. Fill tubing with 5% HCl and let stand for 6-12 hours.
4. Thoroughly rinse the inside of the tubing with deionised/distilled (3.5 times). At a minimum the last rinse should be deionised water.
5. Re-assemble tubing and store in a clean plastic Ziploc® bag labelled with date of cleaning and the initials of the cleaning personnel on the bag. Store and transport the cleaned equipment in a clean vessel, in a clean area.
6. Do not soak the lead weight or the hose clamps in acid.
7. All tubing should be replaced annually.
8. All weights should be rubber coated and any exposed areas should be repaired.

Composite Sample Buckets and 10 L Carboys

Nalgene® – Trace Inorganic Sampling

Follow the procedure described for Cleaning Equipment for Trace Inorganic Sampling.

- Seal with lid or cap.

- Lids for the Nalgene® carboys should be place in a clean Ziploc® bag.

Stainless Steel – General Water Quality Sampling

1. Disinfect with ethanol if used for sampling sewage.
2. Wash with a low residue, non-phosphate detergent (e.g. Contrad®, Neutrad® or Extran®).
3. Rinse thoroughly with deionised/RO water.
4. Store in new, clean Ziploc® plastic bags, and/or cover equipment openings with new Parafilm®. Mark the date of cleaning and the initials of the cleaning personnel on the bag. Store and transport the cleaned equipment in a clean vessel, in a clean area.

Stainless Steel – Trace Organic Sampling

Follow the procedure described for Cleaning Equipment for Trace Inorganic Sampling.

Surface Water Depth Sampler

See USGS (2005) for a protocol to clean/decontaminate depth water quality samplers.

Reference

This protocol was derived from USEPA (1994), Environment Canada (1983), BC-RISC (1997a), USEPA (2000a), Alberta Environment (2002), USEPA (2002) and USGS (2005).

For further reading on the cleaning and decontamination of equipment for surface water quality sampling refer to USGS (2005) and refer to USEPA (2002) for decontamination procedures relating specifically to sediment quality sampling.

4.4 Overview of Health and Safety Requirements for Surface Water and Sediment Quality Sampling

Health and safety of the crew members during the sampling program is an important consideration. To properly conduct the work, the field crew must be experienced with the proposed program and the potential hazards; they must have the appropriate safety and personal protective equipment (PPE) and all necessary training; a detailed job safety analysis must be prepared, which includes very specific emergency response plans; and the crew must be aware of any special safety considerations.

Health and Safety Plans

All field programs must be conducted in accordance with recognized health and safety procedures. All Standard Operating Procedures, as detailed within the Alberta Environment safety manual (Alberta Environment 2005), and/or manual of any environmental consulting company hired to implement the procedures, are to be followed.

Safety Equipment

It is assumed that potential hazards associated with the work have been identified within the respective health and safety document, and the safety equipment and PPE necessary to mitigate those hazards have been detailed.

At a minimum, crews should have communication equipment, survival gear (appropriate to the conditions), and first aid equipment. When working on or in close proximity to water, an approved personal floatation device (PFD) is mandatory. Crew members must also be familiar with the chemicals they may be handling, and must have access to the MSDS sheets. The MSDS sheets will detail handling requirements as well as the PPE necessary to work with the chemicals.

Training

All field crew members should have the necessary training to complete the sampling. This will vary depending on the type of sampling, the method of sampling, and the sampling situation (i.e., water body, watercourse).

At a minimum, all field crew members should possess valid Standard First Aid (Level 1), CPR and Workplace Hazardous Materials Information System (WHMIS) certification, not only to ensure individual and crew safety but also to comply with Workers' Compensation Board regulations. Additional required training could also include Transportation of Dangerous Goods (TDG), small boat safety (as required by the Coast Guard), defensive driving, all-terrain vehicle safety, snowmobile safety, bear awareness, and wilderness first aid.

Job Safety Analysis and Emergency Response Plans

To ensure the safety of field staff, a job safety analysis (JSA) or hazard assessment is a very important component of any workplan. The JSA identifies what work will take place, lists all of the potential hazards that could be encountered during the work, and details the measures necessary to avoid or mitigate the hazards. This includes a list of the PPE required for each component of the field sampling (i.e., driving to the worksite, ATV loading/unloading and access to remote sites, boat loading/unloading and operation, specific sampling methods).

An additional component of the JSA is an emergency response plan (ERP). This document contains all of the necessary information should an emergency situation arise. This includes contact numbers for all persons involved with the project, local emergency contact numbers (EMS, police, fire), hospital contact numbers and evacuation route(s), and any other information necessary for an emergency situation. A component of the ERP is a check-in time where the field crew will check in with a project manager on a daily basis to confirm their safety and the progress of the work. If this contact is not made, the project manager would initiate emergency measures.

Tailgate safety meetings are another important component of a safe field program. These meetings, held daily, detail the work to be completed during the course of the day and allow the opportunity for all crew members to discuss any issues, concerns or improvements to the safety of the sampling program.

Specific Safety Considerations

Safety considerations relevant to specific sampling conditions have been identified below with respect to Lakes and Reservoirs, and Rivers and Streams. This list is by no means comprehensive and all field crew members should refer to a detailed health and safety manual for complete safety considerations and requirements.

Lakes and Reservoirs

Sampling a lake or reservoir will involve either sampling from the shore, or from a boat. Each situation presents special considerations and must be approached in a different manner. Another factor to consider is weather as it could significantly affect sampling, especially from a boat.

When sampling from a lake shore:

- Perform a visual inspection of the substrate in which you will be standing to obtain the sample to ensure there are no obstructions or drops offs that may increase the likelihood of slipping or falling.
- Ensure proper stance and firm footing before obtaining the sample.
- Obtain the sample and return to shore following the same path you walked out on.

- If you feel that the conditions place you at risk of personal injury, DO NOT attempt this task.

When sampling from a boat:

- Perform a visual inspection of your surroundings paying close attention to wave height and direction.
- Ensure that the anchor is secure and the boat is pointed into the wind.
- ALWAYS WEAR YOUR PFD.
- Move within the boat using slow, calculating motions, thereby minimizing risk to oneself as well as others in the boat.
- Do not stand in the boat to obtain the water sample. Position yourself securely on the floor of the boat or on one of the seats.
- Inform others in the boat that you are about to obtain the sample and instruct them to counter balance the boat by positioning themselves on the opposite side to which you will obtain the sample.
- Obtain the sample and return to your regular position in the boat.
- If you feel that the conditions place you at risk of personal injury, DO NOT attempt this task.

Rivers and Streams

Rivers and streams present different considerations from lakes and reservoirs. And the crew members must recognize that conditions within such watercourses can be extremely variable due to river size, depth, substrate, and velocities, among others. When sampling an unknown watercourse (i.e., a watercourse with which one has no prior experience), the field crew must be prepared for the worst case scenario. As with Lake and Reservoirs, sampling can either take place from shore, or from a boat, provided the river is large enough to require it.

When sampling from a watercourse shore:

- Perform a visual inspection of the substrate in which you will be standing to obtain the sample to ensure there are no obstructions or drops offs that may increase the likelihood of slipping or falling.
- Perform a visual inspection upstream to identify possible hazards such as debris that may float down and cause you to lose your footing.
- Perform a visual check downstream to look for obstructions or possible hazards in the event that you are swept in by the current or floating debris.
- Ensure proper stance and firm footing before obtaining the sample.
- Always face upstream or into the current.
- Obtain the sample and walk back to shore following the same path you walked out on, focusing your vision into the current.
- If you feel that the conditions place you at risk of personal injury, DO NOT attempt this task.

Additional points to consider include:

- In high flows, one person, wearing waders and a PFD, samples, while a second person remains on shore with the throw-rope in case of an emergency. (refer to detailed health and safety manual for further details).
- When sampling by helicopter - carefully follow the safety procedures for helicopter sampling (refer to detailed health and safety manual for further details).
- When sampling by watercraft - follow safety procedures for watercraft sampling (refer to detailed health and safety manual for further details).
- Be careful when sampling in turbid rivers - the river bottom can drop off suddenly - use a probe if unsure of the site.
- Be especially careful when working on ice, variables such as ice thickness present a significant hazard and extra precautions must be taken, and additional PPE will be required (i.e., ice testing equipment, harnesses, floater suits) (refer to detailed health and safety manual for further details).
- When river conditions are extremely dangerous (i.e. break-up) take samples from a bridge using a rope and bucket (refer to detailed health and safety manual for further details).

References

The compilation of the preceding safety information was obtained from Alberta Environment (2002), Environment Canada (1983), EMAN-North (2005), North/South Consultants (2005), RAMP (2005), and USGS (2005), and Alberta Environment (2005).

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APPENDIX A Example of a Multiprobe Calibration Record Sheet

UNIT # ZZZZ

DATE OF CALIBRATION : ________________ TECHNOLOGIST : ________________

Post Service Calibration ☐ Pre Field Calibration ☐ Field Calibration ☐

Batttery Voltage :
Surveryor 4 ________ (If voltage is below 7.0 - charge unit) Charged ☐
Scout ________ (If voltage is below 11.5 - change batteries) Changed ☐
Sonde - Internal ________ (Change everytime before deployment) Changed ☐

Temperature : Sonde Temperature C. ☐ ________ Hg Thermometer C. ☐ ________

Dissolved Oxygen :
Barometric Pressure Calibrated @ ____________ mm Hg
Precalibration Reading ________ mg/l ________ % Saturation
Calibrated Reading ________ mg/l ________ % Saturation
Post Field Calibration ________ mg/l ________ % Saturation
(if necessary)

Specific Conductance :
2 Point Calibration For Graphite Probes
(1) Dry Calibration Calibrated to 0.000 Yes ☐
(2) Solution Calibration From ________ To ________
1 Point Calibration For All Other Probes
(1) Solution Calibration From ________ To ________

pH : Minimum of 7-10 minutes **Per Solution** to Stabilize Properly
pH 7.0 Buffer From ________ To ________
pH 10.0 buffer From ________ To ________
or
pH 4.0 Buffer From ________ To ________

Additional Parameters :
Parameter : ________ From ________ To ________
Parameter : ________ From ________ To ________

Internal Logging :
File Name : ______________________________
SDI Address : ________ SDI Delay : ________
Date / Time Set to National Time Signal : Yes ☐ No ☐ (613) 745-1576
Start Date : ____________ Start Time : ____________
End Date : Default ☐ or Set ____________
End Time : Default ☐ or Set ____________
Sensor Warmup : ____________ Circulator Warmup : ____________

Parameters Enabled :
Standard Configuration : ☐
(Temperature, pH, Specific Conductance, D.O % Saturation, Dissolved Oxygen)
Other Configuration : ☐
____

Parameters Order :
Standard Configuration : ☐
(Temperature, pH, Specific Conductance, D.O % Saturation, Dissolved Oxygen)
Other Configuration : ☐
____

CALIBRATION : FAILED ☐ PASSED ☐
(If calibration failed then the unit must be red tagged with full details of problem)

NOTES:

APPENDIX B Example of a Lake Profile Data Sheet

APPENDIX C Reagent Preparation

Group Sample #

Comp Sample #

Project Code

Comp Time

Meter Number

Light Sensor Number

LAKE:

BASIN / CELL:

DATE: Day Month Year

File Name:

Lab #

Comp Code Matrix Type Collection Sampler I.D. Initials

Profile Code Matrix Type Collection Sampler I.D. Initials

Sample #	H H M M	Depth (m)	Light	Temp. (C)	D.O. (mg/L)	Cond. (us/m)	pH	Redox (mv)		
		0.025								

Air Temp (C)

Wind Speed (km/Hr)

Wind Dir.

% Cloud

Waves (m)

Snow Depth (cm)

Ice Depth (cm)

SECCHI (m)

EUPHOTIC DEPTH (m)

BOTTOM DEPTH (m)

QC Sample #	Time	QC Name	Actual Name	Type	Collection

COMMENTS :

Winklers: Corr. Factor : Titrated by :

Location/Depth	Bottle #	Value	Corr. Value	Avg Value

C.1 Acid Wash

1. Safety equipment must be worn when mixing these solutions or when transferring equipment in to or out of the acid bath. This includes rubber gloves and apron and eye protection.
2. ALWAYS ADD ACID TO WATER. Add acid slowly to the deionised/RO water, in a fume hood or well ventilated area.
3. Baths should be changed every four months or whenever contaminants have collected on the bottom.
4. Acid baths are not used for low density plastics, rubber or metal.

C.2 Buffered Formalin Solution

Used to preserve zooplankton and benthic samples.

1. 20 g sodium borate, 1 L 37% formaldehyde, glycerin
2. Mix the sodium borate into the formaldehyde until dissolved. Solution should be prepared at beginning of field season.

C.2.1. Lugol's Solution

Used to preserve phytoplankton samples.

1. 50 g pure iodine (I2), 100 g potassium iodide (KI), 1 L double deionised water (DDW), glacial acetic acid
2. For stock solution: dissolve the KI in ~300 ml DDW in the volumetric flask. Add the I2 , mix well, then top up flask to the mark with deionised water. Wrap flask in foil, date and store in a dark place.
3. For field use: add 10 ml glacial acetic acid to 90 ml stock solution in a small graduated cylinder. Store in a small (~100 ml) glass bottle that has been wrapped with electrical tape to prevent light penetration.
4. This acidified field solution should be made fresh each week. 3 ml of acidified solution are added in the field to each phytoplankton sample.
5. Stock solution should be made fresh every 6 months.

C.2.2. Starch Solution for Winkler Titration

1. Mix 5 g soluble starch with ~50 ml deionised water in a small beaker.
2. Add this suspension to 1 L boiling deionised water and boil for a few minutes.
3. Allow to cool and settle out overnight.
4. Pour off the clear supernatant (if necessary) and store clear solution at 4 °C.

C.2.3 0.025N Sodium Thiosulfate - Titrant for Winkler DO

1. Prepare 0.025N sodium thiosulfate by diluting purchased stock solution, if necessary.
2. Standardize: Prepare the following solution in triplicate without introducing oxygen into standards by proper lab technique:

- dissolve by gently swirling minimum 2 g (~2.1) of KI (potassium iodide) in approx. 100 ml deionised water (Solution #1) in a 250 ml Erlenmeyer flask
- pipet EXACTLY 20 ml potassium bi-iodate solution into solution #1, using a Class A volumetric pipet (be sure to let pipet drain the required amount of time)
- fill flask up to the 200 ml mark with DW and swirl gently to mix
- add 2 ml conc. H2SO4 and swirl gently to mix

3. Titrate the three standards as a normal Winkler sample.
4. Correction factor = 20 avg. mls titrated
5. Standardize this weekly or whenever new solution is added from the stock jug.

C.2.4 1% Saturated Magnesium Carbonate ($MgCO_3$) Solution

1. Add 10 g $MgCO_3$ powder to 1 L volumetric flask. Top up to mark with deionised water.
2. Shake well and fill eyedropper or small squirt bottle for use during chlorophyll filtering.
3. Replace stock solution as necessary, or every six months.

APPENDIX D Specific Remote Procedures: Hydrolab® Datalogger and ISCO® Automated Sampler

D.1 Hydrolab ® Datalogger

Sections C.1.1 and C.1.2. pertain to the datalogging capabilities of the Hydrolab® Series III and Series IV equipment. This section is designed only as an accompaniment of helpful hints and should not be used to learn the procedures of setting up datalogging. It is very important to read the appropriate user's manual to become familiar for the procedures of setting up and capabilities of the Hydrolab® equipment as dataloggers.

D.1.1 Set up procedure

Read and understand the user's manual for the Surveyor III and Surveyor IV dataloggers.

D.1.1.1 Laboratory:

- Make sure that the battery power is fully charged. Connect sonde or H20 to surveyor and ensure all equipment is functioning properly.
- Ensure there is enough battery power to complete the field task.
- Before field deployment, perform a test to ensure the system is operating properly.

D.1.1.2 Manual logging:

- Set the system up making sure the connections are secure. Turn surveyor on and check that clock is set to proper date and time.
- Follow the procedure set out in the user's manual for manually logging data. The Series III equipment only requires an annotation, then by pressing the store button data will be logged. The Series IV surveyor requires a file to be created followed by an annotation so it can be manually stored.
- To download data a PC is required. Connect surveyor to PC and use ProComm Plus or Hyper Terminal to communicate to surveyor. Follow procedures in user's manual for retrieving/transferring files.

D.1.1.3` Self logging (time-triggered)

- Set the system up making sure the connections are secure. Turn surveyor on and check that clock is set to proper date and time.
- Follow the procedures set out in the user's manual for self logging. Both the Series III and the Series IV equipment are similar in the procedures for self logging.
- Secure sonde within water column. Begin datalogging.
- To download data a PC is required. Connect Surveyor to PC and use ProComm Plus or Hyper Terminal to communicate to surveyor. Follow procedures in user's manual for retrieving/transferring files.

D.1.1.4 General

- Surveyors have an internal lithium battery that controls the internal clock. If the Surveyors fail to work, the lithium battery may have to be replaced. Often the display will lock up when the battery fails. The life span of one of these batteries is about 2-3 years. However in the Series III equipment they have lasted up to 5 years.

- It is generally a good idea to check the file being stored while you are logging to see that the data is being logged properly. This can be done at anytime during the logging run and will not affect any datalogging file.
- It is important to download your data immediately upon returning from the field, and once you have downloaded and verified your files to a PC, the file from the surveyor is deleted. This will keep the file directory clear and will make it easier to operate for the next user.
- If you are experiencing difficulties in creating files or logging consult the user's manual or somebody with experience in datalogging.

D.1.1.5 Reference

This method was taken directly from Alberta Environment (2002).

D.1.2 Procedure for the deployment of Hydrolab® equipment

Read and understand the user's manual for the Datasonde III and Datasonde IV.

D.1.2.1 Laboratory

- Replace old C cell or AA batteries with new ones. Connect to a PC and ensure datasonde is working properly.
- Perform two full calibrations on sonde before entering the field. On the day before field deployment, ensure all parameters calibrate properly, and records of the calibrations are entered into the calibration book. On the day of deployment, calibrate and record again.
- Create the logging file as set out in the user's manual. Ensure that file name is correct and date and time are set.
- It is a good idea to wrap duct tape around sonde body, especially where the housing joins are. This helps seal up the unit, adds a little protection and prevents staining on the sonde, making clean up easier.

D.1.2.2 Field use

- Deploy sonde in appropriate sampling area, free of debris and other natural entanglements. If sonde is being used in a river make sure it is placed in an area of adequate flow away from back eddies or areas where it may become buried in silt deposits.
- Anchor sonde protective cage to the site. This is done using 5/16 galvanized aircraft cable from the sonde cage to a stationary object, such as a tree, T-bar or stake, bridge pillar, etc.
- Carefully, place probe guard or external stirrer onto sonde. Place sonde into protective PVC or metal cage. A mesh screen may be used around the probe guard/stirrer in winter for slushy conditions. Using a 0530 Master lock, lock the cage shut.

- Place cage into water column. If cage is laying on river bottom then have the probes facing downstream so that the water flow will cascade over them; if possible, suspend cage into water column.

D.1.2.3 Retrieval

- Pull entire cage from sampling location. Remove sonde from cage and replace probe guard or stirrer with probe cup filled with clean water.
- If sample site is not being used anymore, then clean entire area, removing cage, cable and all debris to minimize impact on environment.
- Download data as set out in user's manual, immediately upon return to lab. Place datasonde on shelf to be cleaned with other sondes. When time permits service sonde.
- If the unit was deployed in or near effluent, rinse with tap water after use.

D.1.2.4 General

- Always have a backup sonde ready in the event that there are discrepancies during calibration.
- Whenever calibrating away from the office, know the barometric pressure for the area and use it when calibrating % sat.
- When deploying during open water seasons make sure the sonde can be retrieved in the event of a flood, and anchored as to not be carried away downstream. Conversely; deploy sonde in deep enough water that in the event of subsiding water levels the sonde will still be in the water column.
- When retrieving sonde in ice covered water bodies be mindful of where sonde is located to avoid damage with pick or chainsaw.
- Always keep a detailed record of maintenance and calibrations of all sondes.
- To minimize bacterial growth, a small amount of Sparkleen brand lab detergent is added to the storage cup water.

D.1.2.5 Reference

This method was taken directly from Alberta Environment (2002).

D.2 ISCO® Automated Sampler Model 3700

D.2.1 Equipment

- ISCO® Sampler Model 3700
- 12 volt marine battery (2 batteries required for heat tape)
- Wiring for battery connections
- Built in Timer to turn on inverter for heat tape at appropriate times
- Solar panel with voltage regulator, if heat tape will be used
- Bag holders with caps
- Disposable bags
- Retaining ring for holders
- Intake tubing PVC 3/8X5/8

- Intake strainer with hose clamps
- 12 volt DC to 110 volt AC inverter
- Foam pipe insulation
- Heat tape (household variety)
- Pump tubing: Silicone 3/8 X5/8
- Plastic tubing connectors

D.2.2 Sampling Procedure

1. For early spring sampling, tape the heat tape to intake line and cover with pipe insulation.
2. Install the intake tubing to required length.
3. Install intake strainer high enough off the bottom that no unnecessary sediment will be collected.
4. Install the inverter close to the ISCO® sampler out of the elements. The ISCO® is connected directly to the battery.
5. Program the ISCO® (see below).
6. Plug in the heat tape to the inverter if necessary. Fasten the solar panel to a secure surface and connect to the battery making sure that positive and negative wires are hooked up to proper terminals on the battery. Note some solar panels have +/- wiring reversed.
7. Set the timer to go off 10-15 min before sample will be taken.
8. Place disposable bags in the holders.
9. Install holders in ISCO® base without the caps, securing the bag to the neck of the bag holder with an elastic band.
10. Install retaining ring.
11. After samples have been collected discard the disposable bags.
12. At the end of sampling season discard pump tubing and intake tubing. Acid wash the strainer. Clean the ISCO® sampler with bleach and water, if mice are present in sampler compartment.

APPENDIX E Example of a River Field Data Sheet

S W — Sample Number

S W — Group Sample Number

A B — Station / Transect Number

A B S — Project Number

Station Descrip/Comment

Sample Date: Day / Month / Year Time MST

Samplers ID1 ID2

2 1 1 — Agency Matrix Type Collection

EndDate: Day / Month / Year Time MST

Sample Depth meters

PROJECT

CREW PRINT NAME INITIAL

FIELD UNIT # WINKLER BUCKET

DATASONDE IN at MST

DATASONDE OUT at MST

Data Download File Name

TYPE OF QA / QC
DETAILS ON BACK PAGE

BLANK ☐
BLANK DETAILS ON SEPARATE SHEET

DISCHARGE MEASUREMENT ☐ METER #

Rating Date Rating Number

DISCHARGE STAFF GAUGE / ECG

MAXXAM Lab #

STATION CODES

Time (MST)

Sample Number S W

GPS LATITUDE

GPS LONGITUDE

100925	**Water Temp °C**			
100923	**pH**			
100924	**Cond uS/cm**			
100922	**Dissolved Oxygen - meter (mg/L)**			
	% Saturation			
	Dissolved Oxygen - Winkler Bottle #'s			
8161	**Winkler DO - Avg. of Titrated Values (mg/L)**			
97060	**Air Temperature C (only measured values)**			
100529	**Bacti / Fecal Coliforms**			
100532	**Bacti / E coli Coliforms**			
99212	**Chlorophyll mg/m3**			
15422	**Total Phosphorus mg / L**			
15114	**Dissolved Phosphorus mg / L**			
	Bacti ID #			

	Absent	Low	Med	High
Turbidity	☐	☐	☐	☐
Color	☐	☐	☐	☐
Foam	☐	☐	☐	☐
Odor	☐	☐	☐	☐
Flow	☐	☐	☐	☐

	Left	Centre	Right
Collected @	☐	☐	☐

	[illegible]	[illegible]	[illegible]
Flow from Prior Visit	☐	☐	☐

Field Preserved

☐
Please Check

☐ Rout ☐ G1 ☐ Nutrient ☐ G2 ☐ CARBON / NC ☐ G2-F ☐ LLN ☐ TOC ☐ DOC ☐ BOD ☐ SULFIDE ☐ CYANIDE

☐ ICPMS Metals (ARCV) ☐ ULTRA TRACE Hg (ARCV) ☐ Low Level Hg (ARCV) ☐ TOTAL Metals (MAXXAM) ☐ DISSOLVED Metals (MAXXAM)

☐ Pesticides ☐ EPP ☐ VPP ☐ RFA ☐ PPCP ☐ PAH ☐ HCE ☐ HCV ☐ AOX ☐ CHLORATE ☐

☐ CHLORO ☐ PHOSHORUS TUBES ☐ Bact. FC / EC ☐ Giardia / Crypto ☐ Turbidity ☐ ☐ Ortho Phos ☐ Isotopes

COMMENTS / OBSERVATIONS Current Weather, Recent weather events, and Unusual Activities, Etc.

PRINT NAME INITIAL

Filtered By

Alberta Environment

ENTERED

FIELD VALIDATED Initials Date
dd / mmm / yyyy

Copy of Fied sheet sent to Biologist ☐

Original Field sheet sent to Biologist with final package ☐

dd / mmm / yyyy

Total River Width (Meters) **103118** ____________

Epilithic Chlorophyll Taken

Distance from Left Bank - m (**97202**)	Distance from Left Bank - % (**97203**)	Replicate #	Time (MST)	Sample Number	McIntyre Lab Values (**101942**)
				S W	
				S W	
				S W	
				S W	
				S W	
				S W	
				S W	
				S W	
				S W	
				S W	

Please Note

% of Algae cover ____________

% Macrophyte cover ____________

Substate Type & % ____________

____

Replicates from each bank ____________

# of rocks per filter ____________

Area scraped sq / cm ____________

QC SAMPLES (PUT BLANKS ON A SEPARATE SHEET)

SAMPLE NUMBER	TIME	FIELD NAME	TYPE	COLLECTION	LAB	Bact ID #
S W						
TP - 15422	TDP - 15114	Chloro - 99212	FC - 100629		EC - 100632	
S W						
TP - 15422	TDP - 15114	Chloro - 99212	FC - 100629		EC - 100632	
S W						
TP - 15422	TDP - 15114	Chloro - 99212	FC - 100629		EC - 100632	
S W						
S W						

Description of QA / QC General Description ____________
Blank water used (and from which lab) ____________
Collected in Intermediate container ____________
Sample Poured off where ____________

DATE TITRATED ____________ CORRECTION FACTOR ____________

TITRATOR PRINT ____________ INITIAL ____________

LOCATION	BOTTLE #	TITRATED VALUE	CORRECTED VALUE	MEAN	COMMENTS	METER VALUE

Revision Number : 1.8 July 05/2004

APPENDIX F Example of Benthic Invertebrate Field Survey Sheet

Athabasca River Winter Zoobenthos Survey

2003

Site: A.R. d/s Grand Rapids

CREW. xxxxxxx

Velocity Meter # WQ-6

H20 FIELD UNIT # 14217

		Hole 1	Hole 2	Hole 3	Hole 4	Hole 5	Hole 6	Hole 7
	Date	26-Mar-03	26-Mar-03	26-Mar-03	26-Mar-03	27-Mar-03	27-Mar-03	27-Mar-03
	Time	17:30	17:40	17:50	18:10	9:45	10:00	10:15
	Latitude	56 ° 22' 30.1"	56 ° 22' 29.4"	56 °22'28.8"	56 °22'28.4"	56 °22'28"	56 °22'28.2"	56 °22'25.5"
	Longitude	112°39'08.6 "	112°39'08.6 "	112°39'08.6 "	112°39'07.9 "	112°39'08.3 "	112°39'08.3 "	112°39'08.0 "
	Water Depth (cm)	86	84	94	90	105	102	90
	Ice Depth (cm)	40	70	75	28	52	74	70
	Water Velocity (rev)	23.00	25.00	16.00	23.00	20.00	19.00	15.00
	Water Velocity (time)	42.40	45.40	44.40	42.10	44.60	43.90	44.20
	Water Velocity (m/s)	0.371	0.377	0.248	0.374	0.308	0.297	0.234
100925	Water Temp °C	-0.34	-0.34	-0.32	-0.32	-0.32	-0.32	-0.32
100923	pH	7.68	7.66	7.67	7.65	7.85	7.50	7.72
100924	Cond uS/cm	500.00	498.00	498.00	499.00	496.00	497.00	497.00
100922	Dissolved Oxygen	12.30	12.19	12.19	12.20	12.53	12.32	12.26
	% Saturation	87.20	86.40	86.30	86.30	88.30	87.30	86.90
	% Boulder	25.00	25.00	50.00	20.00	25.00	25.00	25.00
	% Cobble	25.00	25.00	10.00	50.00	25.00	30.00	15.00
	% Course Gravel	25.00	15.00	25.00	10.00	10.00	15.00	35.00
	Substrate Compactness	Loose	Loose	Loose	Loose	Loose	Loose	Loose
	Epi Choro (mg/m^2)	0.604	51.954	15.883	0.364	27.846	33.974	14.300
	Dry Weight (g/m^2)	88.0u0	28.250	29.333	45.583	40.667	26.333	199.000
	Ash-Free Dry Weight (g/m^2)	11.583	2.333	7.083	10.833	5.083	5.417	11.750
	Comments:	Si/Sa=25 Mushy sediment under surface stones	Si/Sa=35	Si/Sa=15 "Shalely" Clay underneath surface stones	Si/Sa=20	Si/Sa=40	Si/Sa=30 "Shalely"	Si/Sa=25 wood=5%